Expedition Field Techniques

INSECTS

and other terrestrial arthropods

by George C McGavin

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About the author

Dr George C. McGavin is the Assistant Curator of the Hope Entomological Collections in the Oxford University Museum, one of the worlds' largest insect collections, and Lecturer in Zoology at Trinity College, Oxford. He is a member of the Expedition Council and Advisory Committee of the Oxford University Exploration Club. After obtaining an Honours Degree in Zoology at Edinburgh University he moved south to Imperial College, London and the Natural History Museum where he undertook his doctoral studies.

His researches since have taken him from the tropical forests of Papua New Guinea to the jungles and caves of Thailand. Most recently he has been studying the arthropod communities of savanna tree canopies as part of the Society's Mkomazi Ecological Research Programme in northern Tanzania. George has a particular interest in environmental education at all levels, has written many books on bugs and other insects for adults and children and is also known on radio and television. He currently has three insect species named after him and hopes that they survive him.

About the book

I have attempted to gather together essential information on what I consider to be the most useful field techniques appropriate to expeditions. Although mainly concerned with insects, many of the techniques are very appropriate for collecting many other sorts of arthropod, such as spiders. Rather than give an exhaustive bibliography I have picked what I consider to be key references that will allow an easy entry to the available literature. I may have missed out some good techniques and the names and addresses of suppliers of useful equipment so I hope readers will let me know so that future editions can be amended.

Introduction

So, wherever humans have broken ground,
whatever frontiers humans have explored,
they have discovered that they are latecomers,
following in the six-legged footsteps of insects.

May Berenbaum, *Bugs in the System*. 1995

Insects have been around for more than 400 million years and it could be argued that they are the most successful and enduring life form that has ever arisen on this planet. Insects are abundant and ubiquitous. From the poles to the equator, from the surface of the sea to the highest peaks and from deserts to rain forests it is estimated that there are somewhere in the region of $1x10^{18}$ individuals on Earth at any time. Diverse as well as abundant, insects comprise roughly half of the Earth’s one and a half million known species. There are many more species than those to which we have given names and past estimates have been as high as 100 million. The majority view nowadays is that we share the planet with somewhere between 5-15 million species, of which insects will be a sizeable proportion.

Insects are so important to the continued working of the global ecosystems that, as long as the well-being of insects is safeguarded, the Earth should remain habitable for humans (Berenbaum, 1995). This is not overstating the case. As herbivores, predators, parasites and as a food source for countless species, insects are fundamental in all terrestrial and aquatic food chains. Put simply, without insects, global ecosystems would disintegrate. Insects pollinate more than a quarter of a million species of flowering plant. Even from a purely anthropocentric view, without pollinators we would lose one third of all the food we eat. Insects recycle nutrients, enrich soils and dispose of carcasses and dung. Insects provide us with silk, honey, waxes, medicines and dyes. We use them to control pests (mostly other insects) and weeds. Insects have been revered as sacred, celebrated in art and literature and eaten as human food.

Insects are also important for the damage they can do. On average, one fifth of all crops grown around the world is eaten by insects. They carry a large number of plant, animal and human diseases. About one in six human beings world-wide is affected by an insect-borne illness. Leaving aside stings and allergies the list includes plague, typhus, malaria, sleeping sickness, river blindness, Chagas' disease, yellow fever, epidemic typhus, trench fever,

loiasis, filariasis, leishmaniasis, dysentery and typhoid (see Kettle, 1990; Lane and Crosskey, 1993 and Service, 1996). Insects also destroy stored food products, wooden structures, natural materials and fabrics.

For a readable review of the impact insects have on humans read Berenbaum (1995). Some of the major reference texts that deal with insect biology are Chapman (1982), CSIRO (1991), Gullan and Cranston (1994) and Siva-Jothy (1998). Invaluable information on all aspects of biodiversity, its nature and assessment, can be found in Gaston, (1996), UNEP, (1995), Jermy *et al* (1995), Wilson (1988), World Conservation Monitoring Centre (1995) and World Resources Institute (1992 & 1994).

So, studying insects is worthwhile and you will have no trouble in finding them wherever you go. Your discoveries and observations will add to the sum total of knowledge about these significant animals and, for the taxonomists among you, there are new species a plenty. Remember, there are several millions of unknown arthropods out there and the biology of only a relative handful of the named ones is known.

It might be that you have always wanted to visit a particular part of the globe and, whatever entomological study you do, you want to do it there. On the other hand, you may have heard stories of places where strange or rare insects are to be found. Such desires have driven natural scientists and explorers for centuries and I do not want to dampen anyone's enthusiasm. But it is important that you think seriously about where you are going, why you are going and, most important of all, what you can realistically hope to achieve in the time available. It is fair to say that a minimum of 25% of your total expedition time will be completely non-productive due to various problems such as interpersonal tension, ill health (hopefully trivial), weather conditions and all manner of unforeseen obstacles. Thorough planning and preparation are the keys to success.

Section One

PLANNING AND PREPARATION

But Mousie, thou art no thy lane
In proving foresight may be vain
The best laid schemes o' mice an' men
gang aft agley,
An' lea'e us nought but grief and pain,
for promis'd joy!

Robert Burns. *To a mouse on turning her up in her nest with the plough*. November, 1785. Verse VII

" No one should go on an expedition until they've been on one."

Remark attributed to George C. Varley;
Hope Professor at Oxford University 1948 -1978.

An inspiring lecture or television programme has been the starting point for many entomological expeditions. It also often happens that a researcher at a museum or university sees an expedition as an excellent way of obtaining specimens for the institution's collections or for personal research purposes. It would seem an ideal arrangement. The specimens are needed and the members of the expedition are only too glad to have a sound scientific justification to make their venture more worthwhile. While each case must be judged on its own merits and collecting for other people might be warranted, it is not going to be very interesting for you and might not appeal to some funding agencies. All round it will be much better for your expedition and, perhaps, your future career if you build in a significant intellectual component for yourself right at the beginning. Simply handing over an assortment of moths you have collected in the jungles of Borneo, no matter how complete and beautifully prepared they are, is not going to get the mental pulse racing. I am not saying that you should not collect specimens for other people, in fact it is right that you should make as much of a contribution as you can by depositing valuable material in relevant scientific institutions. But at the same time you need to have a personal angle and give yourself the room to develop an expertise of your own. A dozen little-known or even new beetle species donated to a museum is one thing but, coupled with, for instance, a well analysed study of the effects of habitat fragmentation on the ground beetle fauna of the temperate forests of Western Chile, it is an entirely different matter. While it is true that museums and our

understanding of the natural world have benefited greatly from the acquisitive excesses of nineteenth-century collectors, we have moved on. Random or general collecting of material with no clear aim or scientific justification can no longer be considered an acceptable practice and seldom provides much information of real interest. It is essential to have a much more focused aim.

Expeditions tend to be of fairly short duration with two or three months in the field being common. In the case of university students, the summer vacation is usually the only feasible time and this may, to a certain extent, dictate what and where it can be done. So what sorts of projects are possible? Nature reserves of various kinds in many countries of the world lack even the most basic entomological inventory and reserve mangers are generally keen to help with this sort of project. The problem is that the job is probably going to be much too big for a single expedition to tackle. Careful selection of key taxa is one solution. Perhaps, if the area is dominated by freshwater habitats, you might deal with the dragonflies, surface-living bugs or other important elements of the aquatic insect fauna. Another solution might be to concentrate your efforts on the insects of a particular part of the habitat. Microhabitats such as stream-side shingle or mud banks, animal dung, dead wood of all kinds, water in tree-holes or plant leaf axils and leaf litter are all worthy of close investigation. Around half of the rarest beetle species in Europe are confined to dead, dying and rotten wood. In contrast to the more publicised denizens of the vegetation of tropical rain forests, soil-dwelling organisms have received little attention over the years (see Andre *et al.*, 1994). Detailed study of an unique ecotype or plant species might also provide very suitable and worthwhile projects. Pollution, urbanisation, desertification and the loss of natural habitats as a result of human activities are having serious effects on populations of insects around the world. An expedition might seek to quantify some of these effects in a particular location. Insect pests of humans, their domestic animals and crops abound and have good potential for expedition work.

A selection of expeditions with an entomological bias supported by the Society in recent years are shown in Table 1. The Society holds full records of these expeditions on a database and keeps copies of their reports.

Table 1: (where there were multiple aims [§], only the insect work is tabulated).

Location & (RGS report no.)	Taxa studied & methods	Summary of achievements
Alaska: 2932 §	Spruce bark beetle populations: visual inspection of larval galleries.	Incidence of fungal root disease not correlated with bark beetles. Aspect had a significant effect on beetle attack frequency.
Borneo (Kalimantan): 2535	Diversity studies on ants (hand-collecting); damselflies and dragonflies (transect counts) and butterflies (netting and transect counts).	Ant diversity decreased in forest areas logged or farmed. Odonate and butterfly diversity varied between sites.
Borneo (Sabah): 2267	Six projects ranging from territorial behaviour in a damselfly sp. (netting) to vertical stratification in nocturnal insects (UV trapping) and distribution of litter ants (pitfall traps).	Stream aspect determined population size of *Neurobasis chinesis*. Quantification of insect catches at ground level and 10m. Comparative data on ants from logged and primary forest site.
Comoro Islands: 3052	Butterfly diversity: modified transect walk technique.	Recorded status of all endemic species. Compared abundance of *Papilio aristophontes* in different forest types.
Costa Rica: 1598	Insects of bracken: ULV spinning disc insecticide sprayer.	Quantification of bracken insect communities at different altitudes.
Ecuador: 2654 §	Butterfly diversity: bait trapping using urine, dung, rotting fruit and fish.	1,110 species collected from 27 sites (sea level to 4000m) 7-12 spp. possibly new to science.

Madagascar: 2308	Studies on Sunset moths (Uraniidae); hand nets, UV trapping, hand searching.	Survey of host plants and moth distribution in 13 sites. Discovery of larval food plants. New ecological data on some species.
Malaysia: 3039	Diet of cave swiflets: microscopical examination of regurgitated food.	Identification of differences among the diet of different species and between habitats.
Nigeria: 1979	Termite ecology: feeding experiments with wood baits. Collecting moths and beetles: hand nets.	Of 7 spp., a *Microtermes* sp. was most destructive to baits. Description of other arthropods in nests. One collembolan sp. new to science.
Papua New Guinea: 2262	Studies of wild and domiciliary cockroaches: beating hand searching and fish-meal baited cockroach traps. Moth trapping: UV light trap.	Data on the distribution of a new species of domiciliary cockroach. Collection of insects for museums. One new species of stick insect.
Peru (Tambopata): 1664 §	Community ecology of Heliconia leaf curls: hand searching.	28 arthropod spp. discovered in 10 orders. Leaf and ground beetles and bush crickets common.
Philippines (Palawan): 1984	Distribution and ecology of *Halobates hayanus* (a marine water strider); observation, hand net and transect techniques.	New data on feeding, mating, predation and moulting. Populations of *Halovelia* and *Xenobates* described. 1 species new to science.
Scotland (Loch Ness): 1146	Survey of the insect fauna of Loch Ness: various techniques, UV and Malaise traps, sweep and aerial netting.	New distributional data. Some parasitic wasp species new to science.

Sumatra: 2721	Moth collecting: Barnes Wallis Moth Machine (adapted microlight aircraft) plus UV + light trapping.	Aircraft crashed but 3,500 specimens collected from 9 sites by other means.
Trans Africa: 1834 §	Incidence and reproductive success of cat fleas on alternative hosts: domestic animals combed over a bowl of water.	*Ctenocephalides felis* was recorded from dogs, goats, pigs, guinea pigs and chickens. Implications for disease transmission discussed.
Venezuela (Gran Sabana): 1612	Insect diversity and pollination of orchids by euglossine bees: photography, Malaise trapping.	Data on orchid pollinators and foraging behaviour of carpenter bees. Hymenoptera and other taxa collected for specialists in UK and Venezuela.
Vietnam: 2319	Insect biodiversity: sweep and aerial netting (terrestrial taxa), Surber and kick-netting (freshwater taxa) and capture-mark-recapture techniques (butterflies).	Community structure of 328 species of leafhopper analysed. Micro-habitat preference of 14 water bug species. 122 species of butterfly recorded. Distribution and flight behaviour of *Stichopthalma louisa*.
Vietnam: 2704	Hawkmoth and ant diversity: UV and bait trapping respectively. Cicada sound production: tape recordings.	Sphingidae as indicator species. Increased ranges for 15 spp. 2 spp. new to Vietnam. Comparison of ants of scrub and forest habitats. Song analysis of *Platypleura hipla*.

Having decided where you want to go and what you want to do, in broad terms at least, it is now time to start reading/research. You need to be completely familiar with the specific literature and it is always a good idea to consult an expert who will be able to help you with the details of your particular project. Do you know enough about where you are going and what the fauna is like? Are you going at the right or best time of the year for what you want to do? Seasonality has a huge impact on insect populations and good background reading or reliable local knowledge, if available, will avoid embarrassment. Related to your project, you will certainly need to know how much work has been done before and by whom. Literature searching has been made very easy in recent years by the general availability of computerised databases. If you do not have access to these you can still look up the hard copies of publications like *Entomological Abstracts*, *Ecological Abstracts*, *Biological Abstracts* and *Zoological Record*. The latter is particularly useful as it has a systematic section that lists all new species descriptions. By familiarising yourself with work that has already been done you will avoid useless repetition and will focus your plans even further by suggesting specific avenues of investigation. Importantly, it will enable you to contact relevant specialists who you will almost certainly need to consult sooner rather than later.

You will need to contact potential local collaborators and try to recruit them. Students and staff of local or national colleges and universities or any other appropriate bodies should be approached for advice and involvement at the planning stage, not just tacked on at the end to make your endeavour look politically correct for fund raising purposes. Imagine, for a minute, some Brazilian students coming over to study the insect fauna of the Yorkshire Dales or some Tanzanian students planning an expedition to look at the salt marsh communities of the Outer Hebrides. You would expect them to need local expertise, indeed you would probably say that they would not have a hope of achieving much of value if they did not. Similarly, your project will benefit immeasurably from local knowledge, expertise and genuine scientific co-operation, (Trzyna et al., 1996).

Some other pretty important questions to which you will need answers might include: can I bring specimens back to the UK and, if so, do I need export permits? In past years, and with the possible exception of very rare butterflies and big showy species, no-one batted an eyelid about the export of a load of dead bugs and beetles. If you said you had a case full of flies and ants, people were only too glad that you were taking them away. Things have changed, and the whole area of insect collecting has become very complicated. As far as they relate to your specific project you must be

familiar with whatever local and regional regulations or national laws exist. As soon as you can, you must contact the staff of the appropriate scientific section of the relevant Embassy or Consulate and let them know what you want to do. They can put you in touch with all kinds of useful people and will be instrumental in granting research and export permits. Permits and permissions might cost money and take a long time to be processed so, the sooner you find out about them and get things moving, the better. I have known many ventures fail because research and collecting permits were not obtained in good time. Your local collaborators or hosts may also be able to help sort things out at the other end.

The taxonomy of some insect groups might be straightforward and adequately supported by published keys and descriptions but it is certain that you are going to need the services of a specialist at some point. You might only be interested in broad patterns of diversity and identification to family level but, if the success of the project depends on accurate identification of species, then you must think seriously about this before you go (see page 82). There is nothing more soul-destroying than returning from the field after many hard weeks work to be told that your specimens are preserved in the wrong way to allow proper identification. Other common problems are the lack of taxonomists working on a particular group or the fact that your material may be pretty low down on the list of priorities and it could be many years before it ever gets looked at. I am afraid to say that although you might think your efforts are pretty praiseworthy, the experts might not agree. Once you have given specimens over in the hope that they might be identified, it is possible that you might not see them again. Most of these problems can be avoided by proper planning and consultation.

It is also worth thinking about the future value of the specimens or data you have collected. Physical material should be deposited in museum collections with, similar properly prepared and identified specimens going to the appropriate host institution/country. A useful directory of the insect and spider collections of the world has been gathered together by Arnett *et al.* (1993). Users should be aware that the list is not complete as some institutions approached did not return the questionnaire and the staff of those listed may have changed in recent years. I am not going to dictate here whether home or host institutions should have the first pick of material. The important issue here is the long term survival of important material however this can be ensured.

Even the best advance information can be useless due to unexpected changes in local conditions. The late arrival of rains, for instance, might deal

your whole venture a fatal blow. What seemed easy looking at maps in comfort might turn out to be a logistic nightmare. The mark of a good expedition is how flexible you are and how you deal with problems. It is always good to have a fall-back plan of action - and don't forget the powerful force of serendipity in science. Chance always favours the prepared mind and if you cannot think of something else useful to do in a couple of days then perhaps you should have stayed home in the first place. Among the critical things that can go wrong is the loss of key items of equipment *en route* or during your trip. Of course you are insured but that is not going to help much in the middle of nowhere. You cannot take duplicates of everything but think what bits of equipment really are irreplaceable in the field and take spares where you can. Motorised equipment can cause special problems in the field. Make sure that you are capable of simple routine maintenance and repair and that you have basic spares and an extra spark plug. Petrol can be dirty or contaminated with water and modern small petrol-driven engines can be badly affected. It is best to filter all petrol before use but if you do get a blocked jet someone on your team should know how to unblock it using minimal equipment. As with all equipment, test it thoroughly before you go and, if you are using new or unfamiliar techniques, make sure they work and that you understand them.

A lot of basic insect collecting equipment is quite simple and some items can be improvised in the field. You do not, for instance, need to take poles to hold up collecting trays or malaise traps. These can be broom handles bought locally or cut in the field. Spare terylene or nylon netting and cotton to repair damaged nets and traps might be difficult to get and should be taken with you. (see Making your own equipment: page 68).

Forethought must be given to the need for special chemicals. If you are using knockdown techniques (see page 64) insecticide will need to be sent ahead by air cargo. There are numerous regulations concerning the transport of hazardous substances and you will need to find out about these from the carriers you are using, well in advance. It is impracticable to take all but small amounts of alcohol with you and it is usually available in the host country. I have only rarely heard of any problem with carrying small, well-packed quantities of properly labelled chemicals like fungicides and killing agents, such as ethyl acetate, but if in any doubt you should check beforehand. You may well be able to buy suitable alternatives at your destination (see page 73).

Depending on what sort of field work you are hoping to carry out you may need to think about sampling methods and subsequent data analysis in advance. It is always best to collect data with a clear aim or question in mind at the outset and not try to fit questions to the data collected once you've got back. Will you be undertaking qualitative or quantitative sampling? If you need to measure population sizes you can estimate them using marking techniques and capture re-capture methods or various forms of quantitative sampling (Southwood, 1978). If your purpose is to look at patterns of diversity then familiarise yourself with the various sorts of indices that can be used and how best to sample in the field (Magurran, 1988; Samways, 1994). For data handling and statistical analysis see Samuels (1989) or Moore and McCabe (1993).

Section Two

SAFETY IN THE FIELD

Haraka haraka haina baraka

Kiswahili saying:
hurrying does not bring good luck.

Your major responsibility is to come back safely. You know where you are going and what you will be doing. The next thing to do is to assess what the potential risks might be and to take steps to minimise them. It is not the purpose of this book to teach you about the multifarious dangers of bacterial diseases, bad water, poisonous spiders, scorpions, snakes, plants and large, murderous mammals. I expect you to find out about these topics elsewhere. The medical aspects of expeditions are covered in depth by Warrell and Anderson (1997) and I shall only mention some of the commoner nasty insects. Information on disease-carrying mosquitoes, sand flies, black flies, tsetse flies and other harmful insects is plentiful and appropriate preventative and prophylactic measures should be employed. Even if they do not carry disease the bites of insects can be very disruptive to work programmes. Make sure you know what insect problems there are likely to be before you go. For instance, working outside during certain times of the day in parts of the northern hemisphere is made virtually impossible due to the attentions of midges (Diptera: *Culicoides* spp.), horse flies (Diptera: Tabanidae) and other biting insects. Nets, veils and various repellent preparations should be used. Even if you are one of those odd people who never seem to get bitten make sure that you do not have an allergy to any insect repellent products before you go into the field. For extreme conditions, 100% Deet (diethyl toluamide) on exposed skin and a spray of 30-50% Deet on clothing is recommended. Do not forget that Deet dissolves some plastics, especially the ones used to make cameras and sunglasses. Some products claim to be effective against anything while at the same time being gentle and non-irritant but I recommend that you seek informed and independent advice. Wear long sleeved shirts and long trousers. Although hungry mosquitoes can bite through shirts they find it difficult to get through two layers. Wearing a T-shirt under a shirt, even if it is very warm, is a good idea in certain habitats, such as tropical seasonal forests, during the wet season. If undertaking aquatic insect sampling in low latitudes it is essential that you take precautions against schistosomiasis (bilharzia), a group of diseases of humans and other mammals caused by trematode blood flukes belonging to the genus *Schistosoma*. Small, fork-tailed cercariae swim from the flukes' first host, a

snail, to penetrate the skin of the second host, a mammal. In areas where schistosomiasis is known to be prevalent, the use of wellington boots and rubber gloves is obligatory. Information on various aspects of medical entomology can be found in a variety of books, (Kettle, 1990; Lane and Crosskey, 1993; Service, 1996).

Apart from the nasties mentioned above there are some insects that deserve special mention. In tropical regions many species of social wasp and the Africanised or “killer” bees (confined to parts of Central and South America) can pose very serious problems. The simple answer is to try to keep your wits about you as you move through vegetation. Keep away from any nests you come across even if this means backtracking and going round. If you charge off through any habitat flailing a machete about you are likely to come to grief in any case. Africanised honey bee colonies are very much more sensitive to nearby movement than Western honey bee colonies and they are much more aggressive. Despite being careful you might still accidentally damage or come too near a social wasp colony. If you do damage a paper wasp nest by accident, try standing absolutely still. Vespid wasps see movement very clearly and if you run away it is likely that they will get you anyway. Multiple stings, usually around the head and neck will produce painful swelling and the symptoms of shock. People who know themselves to be sensitive to the stings of bees and wasps must carry antihistamine injections or tablets as their medical practitioner advises. Along with social wasps and bees, other members of the Hymenoptera should be treated with respect. Army and driver ant columns (Hymenoptera: Formicidae: Dorylinae) are best avoided as hundreds of large-jawed soldiers swarming up your trouser legs is not a pleasant experience. That they can easily penetrate human flesh is witnessed by the fact that the heads of soldier doryline ants are used as emergency wound sutures. Velvet ants (not ants but a predominantly tropical family, the Mutillidae) are fairly common, small to medium sized ectoparasitic wasps with disproportionately painful stings. Fortunately the ground-running, wingless females have a fairly distinctive appearance (fig.1) and should be easy to avoid.

The bodies of *Paederus cribripunctatus* and related species of rove beetle (Coleoptera: Staphylinidae) often known as Nairobi Eye Beetles, contain a substance called pederin which is toxic and is an irritant to most vertebrates (including man) (fig.1). The beetles, also known as acid flies and whiplash beetles, are quite small and, although they often have bright, warning colours, it is easy to overlook them, especially when collecting them along with lots of other species. The effects of handling them, and then rubbing your eyes

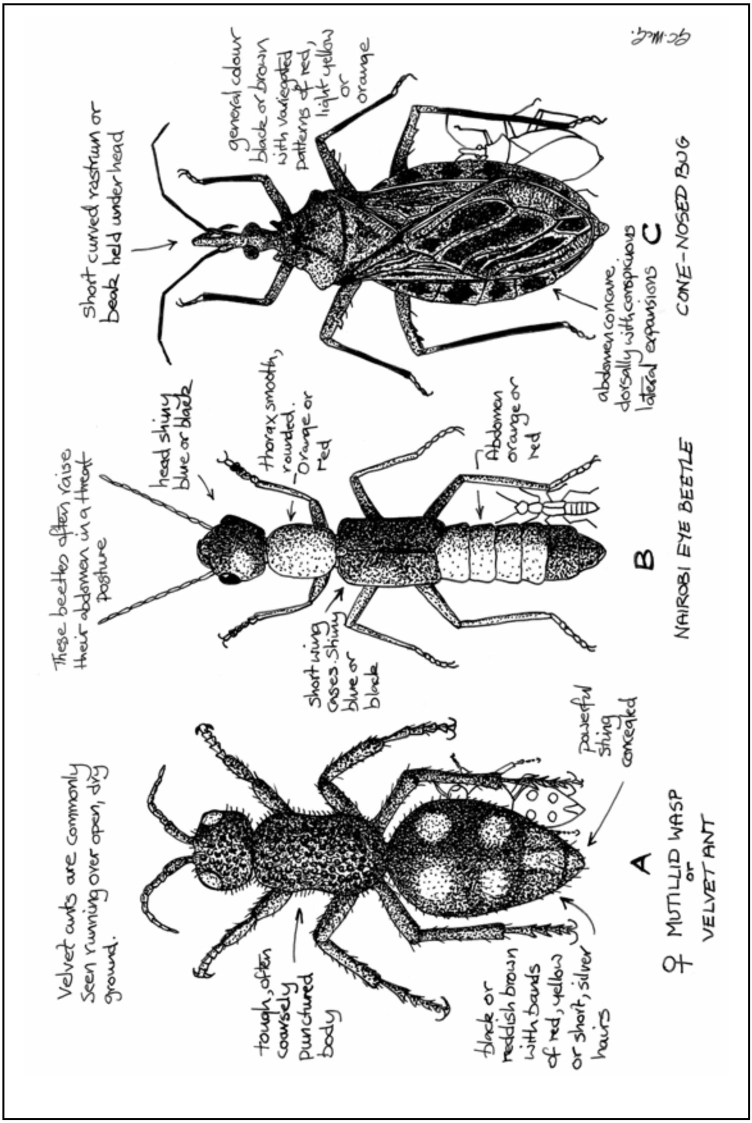


Figure 1: Some insects to avoid

can be very painful indeed and last for days. *Paederus* species occur all over the world, so if you see beetles like these anywhere don't brush them roughly off your skin and, if you do handle them by accident, wash your hands very thoroughly indeed before doing anything else.

Kissing bugs (Hemiptera: Reduviidae: Triatominae) (fig.1) are not so named because they kiss each other, rather they bite sleeping people, especially on the face and lips. The bites can be very painful and cause severe swelling. Triatomine bugs are obligate vertebrate blood-suckers and several common species belonging to the genera *Triatoma*, *Panstrongylus* and *Rhodnius* found in the New World tropics are vectors of *Trypanosoma cruzi*, the protozoan organism that causes Chagas' disease. Some species hide in the crowns of palm trees and, as a general rule, never put your hand anywhere you cannot see (this includes inside collecting nets). Kissing bugs are not the only things that might be lurking.

Walking about in bare feet is not generally a good idea even when off duty. Quite apart from the dangers of sharps and thorns, the sand flea, jigger or chigoe is a significant parasite of humans in dry, sandy areas of tropical Africa and the New World. The tiny females of *Tunga penetrans* (Siphonaptera: Tungidae) burrow into the skin of the foot, often under toenails, where they feed and mature their eggs. The fully distended female can grow to the size of a garden pea. There can be severe itching and inflammation and a number of these fleas can make walking difficult. The associated skin lesions can lead to infection and ulceration which, in serious cases, has resulted in the loss of toes.

Avoid glass tubes and containers if at all possible. They are heavier than plastic and a common cause of accidents. Make sure you know the properties of any chemicals you might be using and follow instructions on their use and disposal (Budavari, 1996). It might be worth taking a small plastic or cardboard sharps bin for the safe disposal of blunt scalpel blades, syringe needles and pins.

As far as tree climbing is concerned, do not do it unless you are fully qualified and trained specifically in tree climbing. If not specified in advance any insurance cover will almost certainly be void. Even if your expedition is visiting a site where canopy walkways and other static structures exist there are dangers and you must consider safety first. If you need to sample from small tree-tops use long-handled pruners or local expertise (they are better at it and probably a lot fitter).



Figure 2: The two people who rolled over in this hired jeep in Thailand were lucky. One had a nasty gash on his lower back which required a minor operation and the other sustained a broken collar bone and concussion

Sampling canopies for invertebrates can be done using a variety of techniques most of which are reviewed by Lowman and Nakarni (1995) (see page 62).

The final word about safety on any expedition must be on driving, whether off- or on-road. Hiring a vehicle with a driver might be an option. If you have to drive make sure the vehicle is fit for the job and road worthy. Cheap vehicle hire might end up being rather expensive. If you are going to drive off-road make sure you get proper training. The bottom line is the simple fact that the vast majority of injuries on expeditions happen as a result of road accidents and not from malevolent beasties. Remember, **25% of all researchers in the field** will be involved in a road traffic accident of some kind (fig.2). You have been warned.

Section Three

CONSERVATION

Innumerable species already belong to the 'living dead',
with populations fallen to levels from which they cannot recover,
even though not all individuals have died yet.

Jared. M Diamond. (1991)
The rise and Fall of the Third Chimpanzee.

Insects should be preserved per se, *so that their activities can maintain ecosystems and also benefit man. Besides, insects, as with other biota, are our earthly companions. Without them we would indeed suffer from a great loneliness of spirit.*

Michael J. Samways (1994)
Insect Conservation Biology.

Whereas once only creatures with fur or feathers were thought worthy of protection, a growing appreciation of the importance of insects has resulted in the protection of many species around the world. Concerns for an ever-increasing number of insect species are being expressed in light of the growing destruction of natural habitats and other environmental changes (Collins and Thomas, 1991; Harrington and Stork, 1995; Samways, 1994). Many countries have national bodies concerned with the conservation of insects and their habitats. Among the influential international organisations that exist, IUCN (the World Conservation Union, formerly the International Union of Conservation of Nature and Natural Resources) based in Gland (Switzerland) and the World Conservation Monitoring Centre based in Cambridge (UK) are particularly relevant for those interested in insects.

Many wildlife bodies have a neutral or ambivalent attitude to non-scientific insect collecting. Insect collecting for its own sake, particularly of butterflies, might be seen as an outdated Victorian pastime but modern collectors who take specimens from the wild and keep them in drawers like stamps or coins will claim that their activities do more to preserve than destroy. It is true that most threatened insect species have become rare primarily due to habitat loss and other environmental factors but the subsequent attentions of unscrupulous collectors has, however, been, and will be the last straw for local or regional populations. The desire for perfect specimens might encourage captive rearing from field-caught individuals.

Excess bred adults can then be released to the wild. I would suggest that this rarely happens and, as with any currency, rarity becomes highly valued. If the collecting community will not police itself effectively then regulations must. But it has also been argued that the protection of some species may make them more of a profitable target. One of the strongest arguments for the continued trade in certain species such as tropical birdwing butterflies is that, properly regulated, it can help to safeguard not only the species concerned but also the habitat that supports them. Semi-captive breeding programmes can provide a significant income for forest inhabitants without endangering natural populations.

Some legislation deserves special mention. CITES, the Convention on International Trade in Endangered Species of Wild Fauna and Flora, lists a few endangered butterflies (*Ornithoptera alexandrae*, *Papilio chikae*, *P. homerus* and *P. hospiton*) under Appendix I (trade completely banned). Appendix II, which allows trade where export permits have been issued, lists *Parnassius apollo*, species of *Bhutanitis*, *Teinopalpus*, *Trogonoptera*, *Troides* and all *Ornithoptera* species (except *O. alexandrae*) (see Collins, 1987). Originally passed in 1900 the Lacey Act was the first Federal law in the United State of America which sought to regulate the internal and international commercial trade in wildlife. As defined at the turn of the century, wildlife meant any wild animal, bird, reptile, mollusc or crustacean and the legislation covered dead bodies, skins, eggs and young. However an amendment to the act passed in 1981 made life very difficult for entomologists. “Wildlife” was redefined to cover the entire animal kingdom and included any part of the species such as blood or even DNA. The Act states that it is a federal offence, punishable by a fine of up to $150,000, to import, export or even cross a State boundary with any wildlife obtained in violation of any foreign, federal, state or Indian tribal law. One of the major problems of the Act is that, in some States, it is being enforced retroactively such that museum collections containing old specimens collected in foreign countries without permits are now breaking the law.

Many countries around the world have passed laws that require the issuing of permits before scientific specimens can be exported. It is your responsibility to find out about them. For instance all dragonflies and damselflies are protected in Austria and Germany. In many countries of Europe it is an offence to collect eggs, caterpillars or adults of any species of butterfly or large moth and, in some parts of Switzerland, you will be arrested for simply carrying a butterfly net. More commonly, protection is extended to particular species. There are several publications that will guide you (Collins, 1987 & 1988; UNEP, 1995). A listing of threatened invertebrates is given by

Wells *et al*. (1983) and swallowtail butterflies by Collins and Morris (1895). The most recent IUCN Red List of Threatened Animals(1996) lists 73 insect species as extinct or extinct in the wild and a further 537 species are recorded as critically endangered (facing an extremely high risk of extinction in the wild in the immediate future), endangered (facing a very high risk of extinction in the wild in the near future) or vulnerable (facing a high risk of extinction in the wild in the medium-term future).

Even once you are sure about the laws that might apply to your project it is best to observe a basic code of good conduct. A code for insect collecting has been set out by the Joint Committee for the Conservation of British Insects/Invertebrates (see Fry and Lonsdale, 1991). A good code of practice for collectors of biological material in general is given on p.57 of Jermy *et al*. (1995).

As a general rule, when working with insects where no laws or regulations apply, or in countries where no protective legislation exists, show much more restraint in collecting large species or those where the taxonomy is well known. You might consider macro-photography as a suitable alternative to collecting (fig.3). Once again get expert advice on your particular situation.



Figure 3: Photographing bees in Tanzania

Section Four

COLLECTING INSECTS

He said, "I look for butterflies
that sleep among the wheat:
I make them into mutton-pies,
and sell them in the street"

Lewis Carroll; *Through the Looking-Glass*
Chapter 8

Insects are largely terrestrial creatures but there are a significant number of species associated with freshwater habitats. There are even some pond skaters (Hemiptera: Gerridae) that spend their lives on the surface of the open oceans hundreds of miles from land. A good account of marine insect species is provided by Cheng (1976).

Collecting and trapping insects to find out what species are present and in what numbers is a basic and necessary part of virtually any sort of field study. The amount of literature on traps of various kinds, particularly those for pest species, is truly immense and available to anyone with access to computer databases or specialist libraries. My intention in writing this book is to provide the reader with the basic information and techniques necessary to carry out insect sampling in the field. Many of the techniques described are also good for catching other sorts of terrestrial and freshwater arthropods and some other invertebrates. For instance, beating foliage, sweep-netting and pitfall trapping will catch spiders just as well as insects. It is worth remembering that if you are studying one particular taxon which you obtain using non-specific or mass collecting techniques you are unavoidably going to end up with a lot of material that you do not want. You should always aim to minimise overkill if you can. If you cannot, then try to find out if someone else might be interested in making use of the material. Good specimens with full data might be of great value to a host country institution whose scientific collections are developing.

Studying plant communities is relatively straightforward. Plants mainly stay still and a quadrat can be used to determine species richness and abundance within a known area. Except for insects such as leafminers and gall formers, the trouble with sampling most insect species is that they are mobile. Methods for collecting insects can be divided into two sorts - **relative** and **absolute**. Relative sampling methods only provide

presence/absence data and perhaps indicate how abundant one species is relative to another. You cannot measure the density of the species (number per metre^2 is the standard measure). Examples of relative sampling methods are pitfall trapping, flight intercept traps and light trapping. In all these cases you do not know what area or unit of the habitat has been sampled. Using absolute sampling methods you are able to calculate the density of the taxa collected. Insecticidal knockdown techniques, suction sampling, hand searching (over a known area) and extraction from a known quantity of leaf litter are all examples of absolute sampling methods. Do not confuse *absolute* with *complete*. No single method will collect all the species present in a sample area but, by using absolute techniques whenever possible, you will be able to make comparisons between different sites or studies.

The indispensable reference work for the study of insects in the field, particularly if you want to measure population sizes, is *Ecological Methods* (Southwood, 1978). Much other useful information on collecting techniques can be found in a number of other publications. For invertebrate collection and preservation techniques (including insects) consult Martin (1977), Lincoln and Sheals (1979), Davies and Stork (1996) and Lott and Eyre (1996). For insect sampling, handling and rearing, see McEwen in Dent (1997) and Powell, Walton and Jervis in Jervis and Kidd (1996). Information specific to aquatic insects groups and flying insects is given by Williams and Feltmate (1992) and Muirhead-Thomson (1991) respectively. Techniques for collecting, rearing and studying immature insects of all kinds can be found in Stehr (1978). An extensive bibliography of mainly British and European work relating to environmental monitoring, surveillance and conservation using invertebrates is provided by Eyre (1996).

Before I go on to describe some of the many and various collecting techniques that you might wish to use on an expedition I would like to make a few general points. Some projects will not require much material to be collected. If you are dealing with aspects of the behaviour or ecology of a single species or small group of taxa you will only need to bring back a very few of each as a voucher specimen to check identifications. Recently many studies of butterfly diversity have employed simple transect walk techniques (see Pollard and Yates, 1993). These approaches obviously rely on the accurate identification of, sometimes very similar, species in the field. If, on the other hand, you wish to do some sort of biodiversity study or carry out a survey of a particular area/habitat/ecotype/plant, you will need to think very carefully how you are going to sample. Total inventories need an incredible amount of effort and you might be better concentrating on keystone taxa (see Gaston, 1996).

4.1 Hand searching techniques

Simply getting down on the ground and looking for insects is a much underrated technique. There are some habitats where no other sampling method will yield results. Imagine you need to look at grass tufts where a particular plant bug lives. Sweeping and suction sampling may not provide much due to the dense growth and, in any case, the foliage might be permanently wet. The bugs in question live right on the surface of the soil and the only way to get them is to go after them with a pooter. A good pooter or aspirator is one of the most useful bits of kit that you can have and designs differ widely (see for instance Martin, 1977). Pooters mostly work with operator sucking, although there are special models for sampling from cadavers and dung which, sensibly, rely on the Bernoulli principle (you blow rather than suck) (Smith, 1986). One of the best, general purpose designs I know of (and have used for many years) uses a Sterilin™ universal specimen tube (approx. 39 x 25mm and 30cc capacity) (fig.4). The tube is made from polystyrene so do not use solvents, such as ethyl acetate, near them. A bung with two holes drilled through it and short lengths of plastic tubing and pipe are all you need to complete the pooter. Insectivores among you should omit a small piece of terylene net secured over the end of the “suck” tube. The diameter of the tubing is important. Too small and you will only be able to collect small species. Too big and you will need a lot of suction to get anything up the tube. Having collected what you want, the pooter tube can be tapped vigorously to shake all the live insects to the bottom and then the bung pulled out and replaced with a screw-top. Large insects can be grasped using a pair of forceps while very delicate insects, if not pootered, can be picked up on the end of an alcohol-moistened paint brush.

Another situation where hand searching is useful is where arthropods have to be sampled from rock surfaces, particularly in caves or lava tubes. In these habitats, populations of animals are often sparse and you can cover a lot of ground with a pooter and a pair of forceps. Different sites can be compared in a quantitative manner by introducing a time element.

The most valuable aspect of hand searching is that you might find out what the animals are doing. Most field collection techniques, such as sweeping or beating, let you know what is present and in what numbers but that is all they tell you.



Figure 4: A pooter designed around a Sterilin™ universal sample tube

Concerns have been expressed about the prolonged use of suction-type pooters such as the one shown here (Douglas, 1984). Although they are simple to make and easy to use there is the risk that minute particles, micro-organisms and fungal spores could be inhaled which might lead to allergies and chest infections. There have been a few recorded cases of small insects invading lungs and sinuses in this way. An improved design of sucking pooter which reduces the danger of inhaling dust, pollen and other fine particulate matter is given by Theron (1985). although they require far less lung power, suck-type pooters should never be used to collect from faeces or carcasses. A blow-type (Bernoulli/venturi effect) pooter for medical, forensic or agricultural work is available from BioQuip® (see suppliers: page 85).



Figure 5: Essential equipment



Figure 6: Other useful bits of kit (PVC tape, gaffer tape, tupperware™ boxes, sorting tray, head-torch, strong lock knife, zip-lock plastic bags, head-band magnifier)

By knowing a little bit of taxonomy you might be able to place specimens into families and thus infer that they were herbivores, predators or parasitoids but you still would not know exactly what they were doing. Night-time searching can be very interesting. Some herbivorous or burrowing insects such as crickets, earwigs and cockroaches may be much more active at night and you might not be able to sample them in any other way. Scorpions are best collected on dark nights when the moon is new or less than half full. Their cuticle fluoresces bright blue in near-ultraviolet light radiating in the range 320-400nm, (Sissom *et al.*, 1990).

Experience has shown that, no matter what you might be doing, a field biologist's kit should contain a number of key items (figs. 5 & 6). Brightly-coloured PVC or marking tape is good for marking plots, twigs etc. Thread, needles, a roll of Gaffer tape (can be used to fix everything from tents and clothes to radiator hoses) and narrow strips of inner tube (buy in the field) will do most field repairs. Do not leave home without fine, permanent marker pens, a Rotring™ pen (or similar), pencils, a Swiss army knife, a couple of pairs of fine watchmaker's forceps, some heavier forceps for coarse work, a selection of fine paintbrushes, a scalpel with spare blades and a fine pair of dissection scissors. Deadwood specialists should take a stout screwdriver or similar implement to prise logs apart. You should always carry a hand lens but, if you are going to do a lot of sorting in the field, you will need a more convenient arrangement. If you have access to a stereobinocular microscope all well and good but they are heavy and not cheap, even if bought second-hand. I have found that a headband magnifier (2-2.5x) is just as good and very much cheaper and lighter. Additional items in your kit might include an old soft toothbrush or two (to brush things off twigs directly into alcohol), a head-torch for night work and a small spray can of household insecticide such as Doom™ or Raid™ in case of emergencies. You will necessarily modify what you take with you in the field to suit your particular project but do talk to the specialists before you go. Their experience will save you time and money.

4.2 Butterfly and aerial nets

Aerial or hand nets are lightweight nets designed for catching flying insects. There are many variants available from any number of suppliers but an excellent net is made by Rani Kasher of the Israel Insect Information Centre (see suppliers: page 85). The net is based on a commercially available telescopic handle fixed to a rim of plastic piping. The net bag is standard fine terylene sewn to a cloth strengthening rim (fig.7). The fully extended handle allows a reach of 1.5 metres (fig.8).



Figure 7: Two aerial nets. A traditional kite-shaped pattern above; Rani Kasher's net below

Nets for catching butterflies specifically should be lightweight and strong with as large a mouth as is practicable. Quick reactions and balletic movements are needed to catch very fast flying or highly manoeuvrable insects such as dragonflies and hesperiids. Some aerial nets are unsuitable because they offer too much wind resistance. Even with a simple item of kit like a butterfly net there are many factors to be considered. Many designs are available so pick one that suits the job or make one yourself. It is difficult to describe the best way to use a hand net as much depends on what and how active your quarry is but, it is as much an art form as Korean ribbon-dancing. It hardly needs saying that fine aerial nets should never be used for sampling insects from dense or thorny vegetation unless you want to spend most of your rest days sewing up holes.

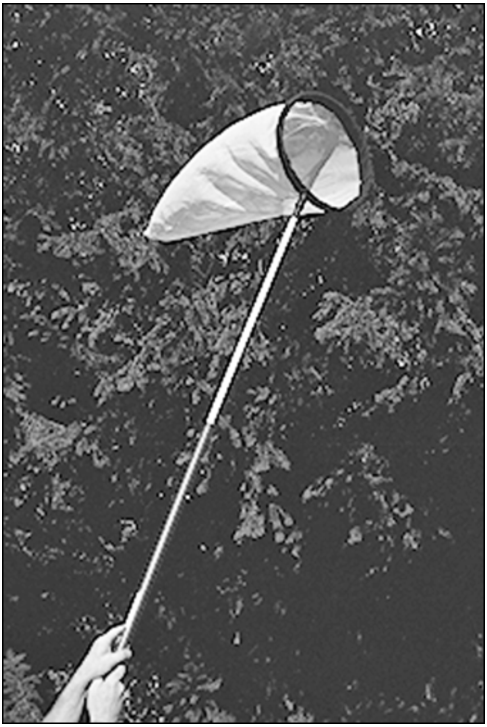


Figure 8: Rani Kasher's net fully extended

4.3 Sweep nets

Sweep nets are made from strong, close-woven material and are designed for thrashing through vegetation. The depth of the bag is important. Some designs are too shallow which allows active insects to escape easily. The bag should be deep enough to allow you to trap the catch by gathering the material together in one hand below the frame (fig.9).



Figure 9: Two sweep nets. The one above is rather shallow and not very wide. The one below is a useful size, has a strengthening bar and it is deep enough so that active insects will not escape too easily

Apart from suction sampling devices (see page 53) there is really no other way to sample insects quickly from low-growing vegetation and grasses (fig.10). Some important relationships between insect diversity, abundance and body size have been revealed using simple sweep netting (Seimann *et al*., 1996).



Figure 10: Sweeping rough grassland

A sweep net is considerably lighter and easier to use than any motorised suction sampler but there are, of course, drawbacks. Sweep sampling is a relative sampling method and is, at best, only semi-quantifiable. You cannot really obtain a meaningful figure for species density. Sweeping will not catch insects that live close to the soil surface and you will miss many of the larger, faster species which, alerted by your vigorous progress through their habitat, will escape before you get to them. Even so you will be amazed at how much material can be collected even from unpromising-looking sites. The sweep net can also be used to collect insects in a rough manner from trees and shrubs. It is impossible to use this technique if the foliage is at all wet as this will result in much of your catch being reduced to a crumpled mush. Keep your eyes out when sweeping in dense or tropical vegetation. Apart from snakes and other things lurking around you might get more inside your net than you bargained for. I have seen a whole paper wasp nest being sampled in a sweep net. The angry buzzing noises from within should be sufficient warning.

To make your results comparable between different sites you will need to standardise your sampling effort in some way. You should use the same net (the mouth diameter of the net is an important factor) and do a fixed number of sweeps per sample (25, 50 or 100 sweeps are typical). How you sweep is important and people vary a great deal in their technique so it is best to get the same people to sample. Another point worth remembering is that you should always sweep into the wind. Describing the proper technique is difficult but keep in your mind that the object is to sweep as evenly as possible from a particular area or volume of vegetation. Stoop down and hold the handle of the net firmly in both hands and sweep the mouth of the net to one side as far as you can comfortably reach. The essential point is that the net should be kept as low to the ground as possible at all times and at the end of each pass the net is turned swiftly to begin the next pass or sweep to the other side. The movement of the net must be nearly horizontal not parabolic. Repeat the sweeping action the required number of times as you walk through the vegetation and, at the end of the last sweep, turn the net mouth or grab the net above the catch so that you do not lose any specimens. The bigger the number of sweeps the more difficult it will be to sort out the catch from the plant debris that you will unavoidably collect as well. If you are sampling for a specific insect species or group you might simply search through the catch *in situ*, collecting what you need with a pooter and returning the rest to the wild (fig.11). Otherwise you are going to have to bag up the material and sort it out back at base. Transferring a sweep net catch to a strong polythene bag is much easier with two people.



Figure 11: Pootering spiders from a grassland sample in Tanzania

One person holds the plastic bag so that the opening assumes the shape of a slit. The other person grasps the net together above the catch. The contents of the sweep net are everted into the plastic bag by gradually drawing the material of the net gradually through and over the retaining hand. Expel most of the air from the plastic bag and tie the top (after putting a label inside). There are two schools of thought as regards to sorting sweep samples (and suction samples) - the live sorters and the dead sorters. The only good reason for killing the catch (for instance with fumes of ethyl acetate) in the field is that some of the catch might start to eat the others. I have never found this to be much of a problem although if there is going to be a long delay between sampling and sorting, killing might be an idea. The reason live sorting is generally preferable is that dead insects, especially small ones, are ten times more difficult to sort out from debris than live ones. If you opt for live sorting try to keep bagged catches out of the sun. Live sorting of sweep samples is greatly facilitated by using a sorting hood.

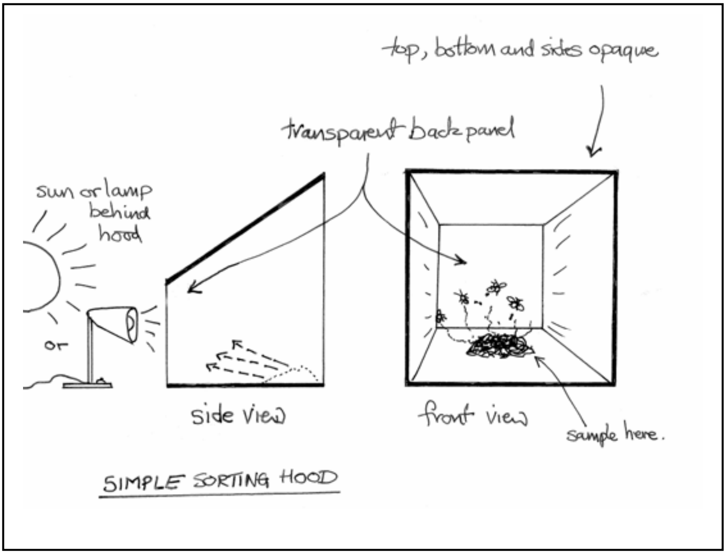


Figure 12: A simple hood for sorting live sweep samples

This simple device, which can readily made up in the field, employs the principle that most insects will move towards a light source. The sides, top and bottom of the hood should be opaque with only the back or part of the back panel transparent (fig.12). With a light placed behind, or the hood simply angled towards the sun, your catch can be carefully tipped out and the live insects pootered or brushed into alcohol as they move towards the light. I have used a cardboard box lined with white paper and a cellophane sheet or fine netting taped to a hole in the back.

4.4 Beating trays

Victorian collectors were smart. You cannot easily collect insects in the rain so why carry a beating tray as well as an umbrella? Although better if made of a white material against which to see your catch, any umbrella held upside-down will serve as a reasonable beating tray.

More specialised designs of beating trays are available from various suppliers and folding designs are easy to take on expeditions. You can make your own with minimal outlay and it is often just as easy to make or improvise something in the field.

The tray is held under the foliage to be sampled and the branch shaken or given sharp, jarring blows with a stick (fig.13). Do not thrash the vegetation as you will damage the plants and end up with a large pile of leaves and debris on your tray which will take longer to search through. Some insects are very good at holding on and you will not, of course, catch internal feeders such as leaf miners and stem borers. Large insects may fly off again very quickly so it is often a good idea to have two people on hand to clear the catch: one to grab the big stuff before they escape and another with a pooter to suck up the smaller specimens. If you are specifically trying to catch very active species a modified beating tray in the form of a funnel-shaped net with a collecting bottle might be preferable. A design called the Copestake tray is commercially available from Marris House Nets (fig.14). It is best to catch spiders and dunk them straight into alcohol in a separate tube as their silk quickly renders a pooter-tube full of insects a useless mess.

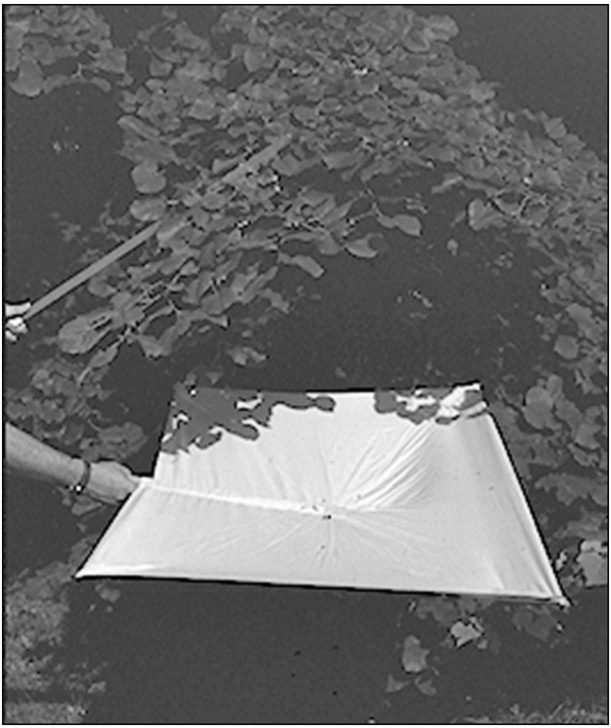


Figure 13: Beating foliage



Figure 14: A Copestake beating trap

Like sweep netting, beating is a very simple and quick technique but also suffers from being rather qualitative. Some studies have claimed that the technique is absolute in that they have beaten over and collected from $1m^2$ trays but it is very difficult to know what amount of the habitat or volume of foliage has actually been sampled. It is also restricted to how high you can comfortably reach. You can, however, try to compare the beaten insect communities from different species of tree or shrub by replicating samples and standardising the effort (the number and vigour of blows perhaps) and area sampled as best you can. The beating of wet foliage is not recommended for the same reasons as sweeping wet vegetation.

4.5 Light traps

That some insects are attracted to light must have been noted shortly after mankind discovered fire. Eighteenth and nineteenth century entomologists used various forms of hurricane lamp and Tilley storm lantern to go “mothing” but in recent decades the use of light sources that produce large amounts of ultra-violet radiation has revolutionised light trapping. There have

been lots of designs of light trap produced in the last 50 years and all exploit the phenomenon that many insects (but by no means all) will fly towards bright lights at night. Moths, notably, are attracted in this way but it should be remembered that it is mainly the males who respond, females preferring to sit in concealment among vegetation. Despite the fact that moths have been collected in this manner for a long time there is still no satisfactory explanation for why they should react as they do. One often-quoted suggestion is that moths use the moon to navigate. If they keep the angle that the light rays subtend to the ommatidia constant they should fly in a more or less straight line. When confronted with a brighter light that is much nearer, the same behaviour will cause them to take a decreasing spiral path into the light source. But not all moths spiral toward a light source; some overshoot, some stop a few metres short and some crash straight into it. There is another big problem with lunar navigation as a biological explanation. Moths are generally not active and will not fly, lights or not, on clear moonlit nights. In fact they are most active and most easily trapped on still nights with heavy cloud cover.

In tropical areas, lights of almost any kind can attract fantastic numbers of beetles, bugs and other insects as well as moths, so much so that, at times, collecting them from a white sheet or painted wall can be an unpleasant experience (a head veil might be useful). Around the world the installation of vapour discharge lamps for street lighting has reduced populations of some moths. Many predators have learned to cash in on this bonanza. Bats will forage in the vicinity of bright lights and birds will clean up the remainder in the morning. In Papua New Guinea I have often observed foraging columns of weaver ants (*Oecophylla smaragdina*) queuing up along lit fluorescent tubes to snatch insects as they fly in to land.

The trouble with light traps is that you cannot be sure what will be attracted to them and what will not, indeed some insects are repelled by lights. Many physiological, behavioural and environmental factors are involved. Many moth species never fly to lights, even though there may be large populations nearby. Perhaps the biggest problem with light trap data is that is very difficult to be sure over what area the trap is effective. In wooded habitats the range of the light may be quite small due to obstructions, but even in open habitats you cannot be sure. Experiments using a 125 watt mercury-vapour lamp have shown that the effective trap radius, far from being the 90 metres originally thought, is rather less than 5 metres.

The best you can do with light traps is to show the presence and perhaps relative abundance of particular species. Quantitative analysis is tricky but, with replications using the same trap design, you might be able to get a rough comparison of different areas or an idea of how the population of some species fluctuates over time. They are thus useful for general survey work but be very careful not to make too much of the data collected.

Of course light trapping has lots of advantages. It is a minimal-effort, crowd-pleaser, can be very productive and exciting and you need only catch and kill (or photograph) the things you specifically want.

Large traps, such as the Robinson MV moth trap which use mercury vapour discharge bulbs, require connection to a mains supply or a generator. This type is not generally useful far from a field station or where you cannot drive a vehicle. Smaller variations of the Robinson trap, such as the Heath Trap that can be run from a 12v car or motorcycle battery, can be used in more remote locations, as long as you can carry the battery. There are a number of portable designs which use smaller batteries permitting their use in very remote or difficult terrain. Various sorts of light trap, their operation and limitations can be found in Southwood (1978), Muirhead-Thomson (1991), Davies and Stork (1996) and Fry and Waring (1996). A very convenient light-trap for use in the tropics is described by Robinson and Jones (1996). The heart of this cheap and simple trap is a 4 watt actinic tube powered by four standard D-size batteries. Larger wattage tubes are available but would need the use of bulkier batteries.

Material that comes to light can be collected by hand or by pooter and should be killed and preserved in a manner appropriate to the taxon in question. Most non-lepidopterans can be put into an ethyl acetate or cyanide killing jar (See Killing Methods, page 71) before being decanted into paper envelopes and stored dried or fresh in a sealed plastic tub with chlorocresol (see Specimen Preservation, page 75). Of course material can be mounted or pinned in the field but this can be a slow process and you might not have the energy or time to do it immediately. Set material also takes up a lot of room in luggage. Despite these drawbacks many workers consider that this is the best course of action for moths. Small or micro-moths should be taken from the killing jar and pinned using a minute or micro-pin vertically through the thorax. They are then pinned into small plastic boxes lined with Plastazote™ (or similar expanded polyethylene foam) and the wings should be set forward and held in place with additional micro-pins. In this manner a large number of fragile specimens can be kept and transported safely. Specialist information on collecting and studying microlepidoptera is given by Sokoloff (1980). Butterflies and robust moths, such as hawk moths, can be papered or

put in small envelopes inside a sealed plastic tub with cholorcresol as an antifungal agent. My own experience with the chlorocresol storage method shows that even delicate butterflies and moths can be spread and set at home months after the end of a field trip (see Field Preservation, page 75). Non-robust moths are best pinned through the thorax with an appropriately sized, stainless steel pin and placed directly in a storage box. There is no need to set the specimens as they can be relaxed and set properly at home. The main advantage of this technique is that the specimens will not get rubbed and lose their wing scales.

Like most skills, the art of setting butterflies and moths is best picked up by watching a professional do it but instructions are given in a number of publications (Arnett, 1985; Dickson, 1992; Martin, 1977; Walker and Crosby, 1988).

4.6 Flight intercept traps

Any sort of static trap which catches insects in flight could reasonably be called a flight intercept trap (FIT). Nowadays the term is used more specifically to describe a single, rather simple type of trap. A piece of material (usually black terylene) is stretched tightly between poles or trees and plastic containers are arranged on the ground along its length (fig.15). The containers (rectangular ice cream tubs will serve well but excellent light, plastic trays are available from GB Nets) are part-filled with water and a little detergent to break the surface tension. Insects flying into a FIT will either crawl up and escape from the top or simply drop downwards. Many other things will just fly off again. Peter Hammond of the Natural History Museum recommends a roof over the FIT and the use of a few crystals of chloral hydrate in the water containers to prevent decomposition of the catch (Davies and Stork, 1996). Apart from not catching a significant proportion of the things intercepted, one of the problems with all flight intercept traps is that you cannot be sure whether they are acting purely passively or if some insects are actively attracted to them. Also if you require directional data, such as whether or not insects are flying in a particular direction, such as in or out of a woodland or other habitat, you will need to make modifications to available designs or make you own. Basic and directional FIT’s are available from Marris House Nets. Insects collected from the tubs can be strained out using a tea strainer or small sieve and transferred to alcohol in labelled tubes.



Figure 15: A flight intercept trap

Flight intercept traps of a particular design are called Malaise traps after an entomologist, Rene Malaise, who noticed that insects entering his tent always seemed to accumulate "*at the ceiling-corners in vain efforts to escape at that place without paying any attention to the open tent-door*". His design, which was published in 1937, has spawned many variants over the years (see Muirhead-Thomson, 1991 and Southwood, 1978 for review). Malaise commented on the main advantages of his trap: it is *"better than a man with a net"* and *"it can catch all the time, by night as well as by day, and never be forced to quit catching when it was best because dinner-time was at hand"*.

Malaise traps are essentially open-sided tents made of material and which have a high point in the roof where the insects tend to collect and from where they are funnelled into a container (fig.16). To increase the efficiency of the Malaise trap, water-filled containers can be arranged on the ground under the central partition. In this way insects that do not crawl upwards in response to flight interruption will also be trapped. There is no doubt that they are effective in catching flying insects and, if lashed to a light frame, they can even be pulled up into forest canopies. Basset (1988) describes an ingenious composite trap based on the principles of a Malaise and a window trap for sampling arthropods in tree canopies. It is often said that Malaise-type traps

are non-attractant but anyone who has used them will know that this is not really true. When working in tropical dry forest with a mainly black malaise trap I was astonished to find, day after day, that the collecting bottle was almost entirely filled with horse flies (Diptera: Tabanidae). Indeed the phenomenon of attraction to large, dark objects has been put to good use in the design of traps for blood-feeding flies. Malaise traps made by Marris House Nets can be obtained made with black walls and white roofs, all black or all white. The three patterns are requested by entomologists around the world in the ratio 10:4:1 (R.S. George, pers. comm.). It is clear from experiments using the three patterns in the same habitat that insects are differentially attracted (Usherwood and McGavin, unpublished results).



Figure 16: A malaise trap set up on a hillside in Tanzania during the RGS's Mkomazi Ecological Research Project

For the majority of survey-type work the collecting bottle of the Malaise trap is filled one third to one half full with 70% alcohol which serves to kill and preserve the catch. If, however, you are specifically collecting Diptera, most species are best collected dry with a killing agent such as a piece of Vapona Fly Killer™ or similar domestic, slow-release strip insect killer containing dichlorvos wrapped loosely in tissue at the bottom of the collecting bottle. When servicing traps distant from your field base remember to take lids, spare bottles and enough alcohol for refilling.

Specific flight intercept variants are sticky traps and window traps. Sticky traps are simply pieces of perspex or glass coated with sticky material (i.e. Hyvis™) and fixed to a post. Window traps should be as transparent as possible and equipped to catch insects that either fall, or crawl upwards to get around the obstacle. Glass sticky or window traps are heavy and pose obvious dangers in the field. The major problem with sticky traps is that the glue used to trap the catch is often difficult to remove and will almost certainly require the use of dangerous and/or inflammable solvents. Even if you get the catch off the traps in one piece it is often very difficult to identify specimens. Sticky traps can be useful for the regular monitoring populations of pest insects where identification does not pose a problem but are not recommended for surveys in the field.

The type of FIT to employ for survey or collecting work will be dependent on what you want to catch and the kind of habitat you are working in. However, a good Malaise design remains a popular choice. If in doubt seek expert advice.

4.7 Pitfall traps

Pitfall traps are a very easy and cheap way to catch active ground-living arthropods of all kinds. They may seem too simple to require more explanation but I have seen enough badly set ones that I shall risk being boring. A hole is dug with a trowel and a plastic container of some kind is sunk into the ground so that its rim is level or slightly below that of the surrounding ground. It is no good at all if there is even the slightest lip showing above ground for small animals will simply walk around the circumference. Bits of debris and foliage that lie across the top of the trap will act like little walkways. Once properly set, pitfalls are left and animals will fall in and the steep, slippery sides should ensure that most things will not escape.

Pitfall traps are usually one-third filled with water or alcohol to trap and kill the catch. Very small insects can sometimes escape from the surface of water so a few drops of detergent are added to lower the surface tension. When using water the catch will start to decay in a day or two, faster in warm weather, so it is vital that the traps are emptied daily. If you are not able to service the traps regularly, or if you wish to leave them undisturbed for a week, you should use a mixture of 40% ethylene glycol (antifreeze) in water as a preservative (but see Holopainen, 1990). Whichever technique you employ the contents of the traps should be preserved as soon as possible in 70-80% alcohol. Of course alcohol can be used in the traps in place of water but it is expensive, will evaporate faster than water and a few studies have suggested that the smell of alcohol can act as a repellent for some species. I do not recommend the use of formaldehyde solutions in arthropod-trapping pitfalls as it is unnecessary, toxic, a suspected carcinogen and can be an environmental hazard if not disposed of carefully.

Pitfall traps can be used dry for live collection of specimens in which case they need to be serviced much more regularly than those where the catch is killed in fluid. For instance, a large ground beetle having fallen into a dry trap will proceed to eat anything else that happens along.

There are many pitfalls in using pitfalls. Sometimes small mammals and amphibians get trapped and die unnecessarily. When pitfalling in dry areas birds and other animals soon learn where to get a drink. Pitfall traps with a large surface area might act as a water or pan trap and attract flying insects as well. It would be impossible without watching it all the time to be sure whether an insect stumbled and fell into the trap as intended or flew into it. Obviously if you find butterflies in your pitfalls it is safe to assume they did not fall in by chance. With large or long term traps it is often a good idea to arrange some sort of solid cover to keep rain out or mesh of various grades to exclude certain sizes of animal. Another problem is that you can never be sure whether even ground-living species might be attracted to them in some way, but as with light trapping, the biggest problem associated with pitfalls is that you cannot be sure over what area they are trapping. Despite these very serious drawbacks, pitfall trapping remains a standard arthropod sampling technique, mainly because it is cheap and produces large amounts of material with relatively little effort (see references in Lott and Eyre, 1996).

A well tried and tested protocol for pitfall trapping any given area employs cheap, plastic drinks cups with a rim diameter of 7cm (internal volume 200-250ml).

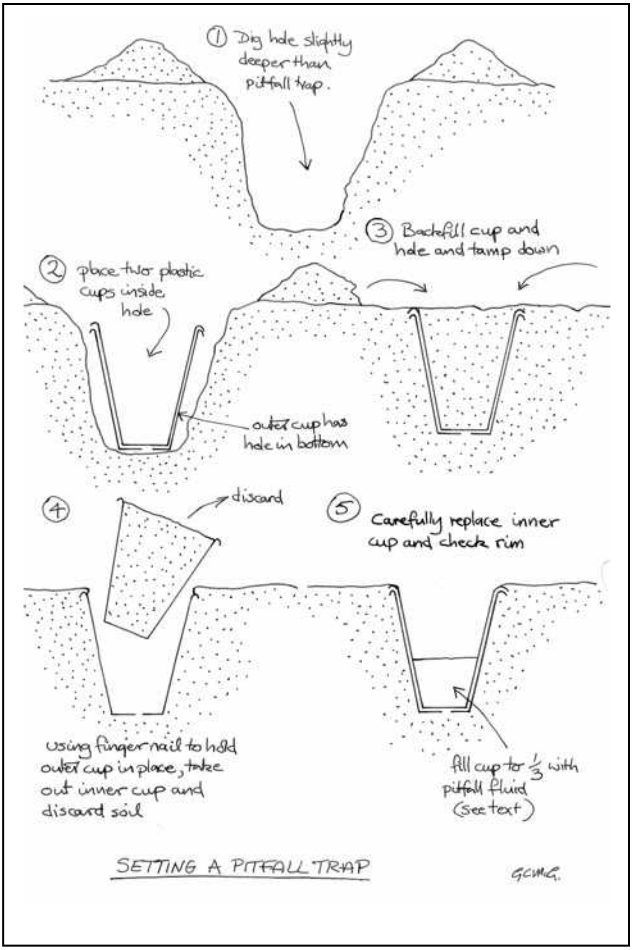


Figure 17: Setting a pitfall trap using plastic drinks cups

The standard unit of trapping effort could be, say 180 trap/days. This could be 60 traps operated over a period of 3 days or 30 traps over 6 days. Traps are placed in lines of 10 with a spacing of 4-5 metres between each trap and between adjacent rows. Each trap unit consists of two drinks cups,

an outer one and inner one (see fig.17). To set each trap, dig a hole larger than the drinks cups with a trowel and make sure it is just deeper than the rim of the inner cup. Put two cups inside each other into the hole and back fill the space around the outer cup. Gently press down and smooth the soil around the rim of the inner cup. Do not worry if the inner cup is now partly filled with soil at this stage. The whole point of having two cups is that you can now take the inner one out and empty excess soil, preferably off-site. The inner cup is now replaced with a clean one. The outer cup protects the inner one and allows subsequent rapid emptying of the inner cup. The outer cup should have a small hole punched in the bottom to allow rain to drain away. If this is not done, rain can seep between the two cups and the inner one will float up, spilling its contents. If you are going to leave the traps for more than a day or two or if there is the likelihood of very heavy rain, it is a good idea to make a neat hole in the inner cup with a paper punch, about 2cm from the rim and glue a small piece of terylene or similar material over the hole. In this way, if the cup fills up, the excess water will drain away and the catch will be retained.

When all the traps are dug in, the inner cups are one-third filled with water containing a few drops of detergent. To save time and depending on the nature of the habitat, the position of the grid can be marked with a cane bearing brightly-coloured PVC tape. Catches from all the traps from one site and one day are collected by pouring them through a sieve and bulked together in 70% alcohol (fig.18). If you really want to keep them, any vertebrates collected should be stored separately from the invertebrate material. As with keeping any biological material in tubes of alcohol, the total volume of the catch should never be more than half the volume of the storage tube. If you completely fill the tube there will be little room for the alcohol which will become diluted anyway, resulting in the decomposition of the catch.

If you want to trap specific groups of insects, such as those attracted to carrion and dung, it is a simple matter to incorporate an appropriate bait (see Bait Traps: page 51). You will of course still catch things that just blunder in but, with experience, these can be ignored. In two years study at six main sites in the Serengeti ecosystem, more than half a million beetles comprising at least 105 species of Scarabaeinae, were trapped in pitfalls baited with different types of dung. (Foster, 1993). The traps consisted of a 5 litre straight-sided, plastic bucket (mouth diameter of 21.5cm) and fitted with a removable aluminium funnel. The traps were dug in flush with the surface of the ground and a piece of wire netting (5cm mesh) placed on the top. The traps were then baited with a measured amount of dung wrapped in mosquito netting. So many beetles were attracted that Foster had to modify the trap by adding a 4 litre paint can to increase the collecting volume (fig.19). Using



Figure 18: Servicing pitfall traps. The catch from the inner cup is emptied into a container and the inner cup refilled with pitfall fluid

simple techniques such as this, he was able to reveal detailed patterns of beetle occurrence,density, competition and dung use. When using carrion or dung baits you will often attract much larger four-legged animals who may eat the baits and destroy the traps so it is important to protect them with strong wire or other exclosures. Foster's design incorporated a cylinder of weld mesh wire (5cm mesh) to minimise disturbance by jackals.

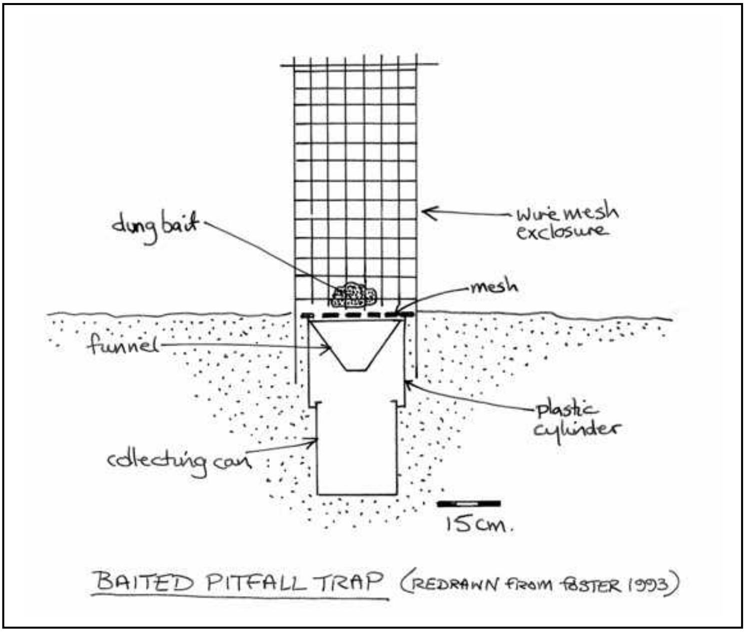


Figure 19: Foster's dung-baited pitfall trap

Recently, the use of subterranean pitfalls traps has produced some very interesting data on the occurrence, abundance and distribution of species that spend most or part of their lives underground. Owen (1995, 1997) describes the construction and use of a simple pitfall trap for the repetitive sampling of hypogean faunas.

Techniques on collecting, trapping and preserving insects from cadavers can be found in Smith (1986).

4.8 Emergence traps and rearing

Collecting insects by trapping them as they emerge, usually as adults from pupae or cocoons following larval development, diapause, aestivation or over-wintering, may be essential if you are going to do anything more than compile a simple species list. Drawbacks are that you might not know how long parts of the lifecycle might take and any material collected in this way will not give a complete picture, however the same could be said of any technique used during an expedition of fixed duration. For some species there may be no other practical collection method. Rearing here does not mean

captive rearing and breeding but rather the means by which adult insects may be collected from leaf mines and stems, from under bark, from dead wood, from parasitised insects, from corpses, from macro-fungi, from the soil and so on. Different sorts of trap will be needed for each habitat/microhabitat. At its simplest an emergence trap or rearing container may be a cotton wool-plugged or muslin-covered tube or tub. Collecting insects in this way will provide much more useful biological information than general collecting techniques. For instance, beating the foliage of an oak tree may yield many species of gall wasp but galls placed inside simple, muslin-covered containers will yield the gall causers, other gall occupants and their parasites, allowing the elucidation of associations and relationships. It will also be possible to associate and preserve larval and pupal exuviae (shed skins) with the emerging adults. Deeming (1993) gives a good account of the simple techniques he uses to rear shoot flies belonging to several families. The same techniques could be equally well applied to the rearing of other insect groups.

By the very nature of their biology, parasitoids collected in isolation from their hosts provide little useful biological information. Powell, Walton and Jervis in Jervis and Kidd (1996) and Shaw (1997) provide information on rearing and studying parasitic Hymenoptera.

Many insect species emerging from soil or litter at the beginning of the growing season are positively phototactic. Emergence traps are easily made from any suitable upturned opaque, usually black, container dug into the substrate and fitted with a transparent collecting bottle at the apex. Such devices, which can also be adapted for collecting insects emerging from or foraging on live trunks or fallen timber, are sometimes called photo-eclectors (see Basset *et al.*, Chapter 2 in Stork *et al.*, 1997). Insects emerging from aquatic habitats can also be collected in emergence traps. Details of several designs of terrestrial and aquatic emergence trap can be found in Southwood (1978), Williams and Feltmate (1992) and Davies and Stork (1996).

Emergence traps can be considered an absolute collecting method provided you sample from a known area or volume of substrate or habitat. Making simple and effective emergence traps to suit your particular purpose may be preferable (and certainly cheaper) to buying a ready-made design. A large, general purpose model often used for processing large amounts of wood, litter, flood debris, compost etc. is the Owen Emergence Trap (see Equipment suppliers, page 84). Measured quantities of material are placed inside the trap and left. Emerging insects should eventually find their way to the collecting bottle at the top. Some traps containing rotting wood have continued to yield material for up to two years from setting up. Other

commercial designs for trapping insects as they emerge from live or dead tree trunks, litter or soil are available from ecoTech.

4.9 Water pan traps

A very simple type of attractive trap can be made from almost any sort of shallow tray or container (usually made of plastic or heavy aluminium foil). The pan is partly filled with water to which a few drops of detergent have been added to reduce the surface tension. You could use an alcohol solution or other preservative but this might attract different species. If you use this technique, be consistent across samples and sites. A bright yellow background colour has been found to be the most effective in attracting small flies, wasps and bugs such as aphids. There are many advantages of this sort of trap. You can put out scores of them very cheaply and you can use them virtually anywhere you can reach from ground level to the tops of trees. The main disadvantage, of course, is that the method is not absolute. As heavy rain may wash out your catches or they might dry up in very hot conditions, they need to be emptied daily or more frequently.

4.10 Lures and Baited traps

To locate food, mates or egg-laying sites insects will respond to all manner of environmental cues. Rather than employing general collecting techniques, specific insects can be attracted to various baits depending on their functional ecology. Indeed many insects will only be caught using this sort of approach. Baits can be divided into major groups: dung, carrion, rotting or fermenting plant material, sexual odours (pheromones) and live animals. There is plenty of scope for originality and ingenuity in the design and refinement of baited traps (fig.20). Southwood (1978) reviews bait trap techniques in general and for blood-sucking flies in particular. A wealth of excellent information can be found in Muirhead-Thomson (1991) who deals with the response of plant pests to visual and olfactory baits and the response of blood-sucking flies to visual traps, live animals and their odours.

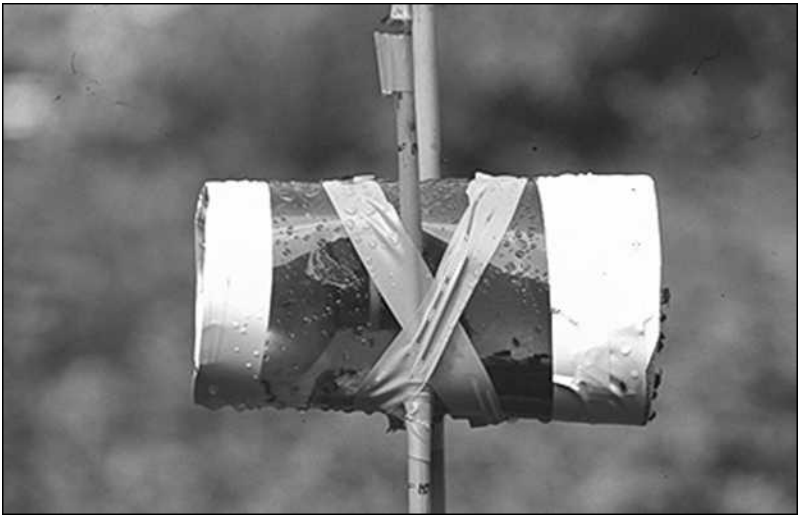


Figure 20: A very simple bottle trap made from a large soft drink bottle, baited with fish. The top of the bottle is cut off and reversed to act like the opening of a lobster pot. The bottom is removed and replaced with muslin to allow a good flow through of air

Animal dung and carrion will attract specialist flies and beetles and these baits can be incorporated into pitfall or flight intercept traps. The attraction of butterflies to putrid fluids is well known. A small dustbin or bucket partly filled with liquefying rotting fruit (add yeast to accelerate process) and/or carrion will attract certain nymphalid butterflies, particularly those of the genus *Charaxes*. A woodland species found across Europe and Asia to Japan, the Purple Emperor Butterfly, was often caught by using "the juices of a dead cat, stoat or rabbit" or "a seething mass of pig dung." Males (mainly) of many species around the world also indulge in puddling. Large numbers of several species may gather to drink around the edge of puddles, urine patches, salt runs and other damp areas. The physiological need for salt and other minerals has been put forward as an explanation for this phenomenon. In tropical regions certain butterflies will also drink from sweat-soaked clothing but there are also species of so-called vampire moth (Lepidoptera:

Noctuidae) who can pierce human skin to drink blood. These interesting insects almost certainly evolved from fruit-piercing ancestors. Butterflies of the neotropical genus *Morpho* can be attracted to lures of bright flashing colour. Materials that reflect ultraviolet will be attractive to many sorts of insects including water beetles.

The technique of "sugaring", now not as fashionable as it once was, relied on the attraction of moths to sweet and fermenting fluids. Old books had many exotic recipes but a good basic mixture would be a thick mush of rotting bananas, brown sugar and dark beer boiled for ten minutes to which is added a splash of rum or essence of pear drops (amyl acetate). The mixture is daubed on posts and tree trunks after dark. The mixture can also be soaked on to thick ropes which are hung up. A head torch is useful when collecting the catch.

4.11 Suction sampling

From the 1950's onwards the need for more quantitative methods of sampling insects led to the development of a number of suction devices (Southwood, 1978). Some of you may have been unfortunate enough to have used a petrol-engine driven D-Vac, or, more correctly, Dietrick suction insect sampler. Originally developed for use in agricultural crop systems, the D-Vac became widely used in ecological entomology and virtually the regulation piece of equipment for sampling invertebrates in grassland habitats. The advantages of using a standard machine with a known sampling area and sampling effort were that different sites and different studies could be compared in a way that had previously been impossible. The disadvantages, however, were many and varied. There was a tendency, among some users, to assume that everything was caught. Machines were, in fact, very variable in their efficiency and few calibration studies were ever carried out. Good practice involved D-Vac-ing for a number of timed sucks combined with careful hand searching afterwards to ensure that nothing had been missed. D-Vacs were very expensive, never very easy or comfortable to use and needed constant maintenance. Despite modifications in the 1960's these D-vacs are no longer available commercially. A very similar machine called the ecoVAC is made by ecoTech in Germany (see Suppliers: page 85). The advantages of suction sampling remain the same and some devices are still available. A smaller, backpack two-stroke, machine called the Univac portable suction sampler™ and another type called the Vortis insect suction sampler™ are made by Burkard Manufacturing Co. Ltd. (see Suppliers: page 84). These two machines are almost certainly too expensive for the average expedition being more than £1700 and £1400 respectively at the time of

writing. The Vortis sampler, despite its high cost does represent a very significant improvement in suction sampling and has many advantages over previous designs (Arnold, 1994). The machine is lightweight, can be simply used by one person and, as insects are sampled directly into a container, the losses when bag emptying are minimised.

The cost of commercial machines has resulted in a number of people making very simple modifications to leaf blowers and similar items of garden machinery (Wilson *et al*, 1993; Stewart & Wright, 1995). Studies have shown that these home-made samplers do a very good job indeed but, like all other designs, they will not sample everything with equal efficiency. You must calibrate your sampling in some way to estimate what percentage of organisms you are collecting. This percentage will vary greatly with insect body size and the sort of habitat you are studying.

There are many inexpensive models on the market but a Sabre BLV 25 Blow-Vac™ costs well under £100 at the time of writing and is incredibly lightweight (fig.21). All you need to do is to make a net that fits neatly inside the suction tube and secure it with a large rubber band. Should the rubber band slip off or break it is advisable to fit a piece of metal mesh to stop the net getting sucked into the fan blades. Work out the cross-sectional area of the suction tube and you will be able to simply calculate the area you have sampled. For low vegetation a sampling unit might be 25 random sucks pooled from within a given area of the habitat. Alternatively you could use a cylinder of known area to isolate the vegetation to be bug-vacuumed. A good technique is describe by Stewart and Wright (1995). Samples can be everted into plastic bags as described in the section on sweep nets.

4.12 Extraction techniques

Separating small invertebrates from the substrate can be a difficult and time-consuming process but scores heavily on being an absolute method. Measured volumes, weights or areas of sample will yield quantitative data. Different techniques are needed for different substrates but the simplest form of extraction tool is a sieve (fig.22).



Figure 21: Suction sampling using a modified garden blow-vac.

Here gravity and agitation will only effect a partial separation. Different gauges of sieve (e.g. 10, 5 and 2mm meshes) might help, but animals may hold on to or hide inside large debris and very small specimens will still need to be separated from fine debris. It is generally better to persuade the animals to leave the substrate under their own steam and this principle is employed in a variety of extraction devices. The obvious advantage is that the samples obtained are relatively clean and require little further processing.



Figure 22: Sieving leaf litter over a tray for cockroaches and beetles in The Gambia

A disadvantage, for some expeditions at least, is that the equipment can sometimes be bulky and, in cases where a heat or light source is required, difficult to arrange in the field. It is worth bearing in mind that, the smaller and more delicate the animals of interest are, the more difficult it will be to extract them efficiently.

Extraction funnels such as Berlese/Tullgren designs are suitable for the extraction of insects and other moderately robust specimens from leaf litter, crumbled soil, decaying wood or even fresh foliage. There are very many variations of the same theme. The measured sample is placed on a mesh or sieve over a funnel which leads to a collecting bottle containing alcohol (or dry for live collection). A (preferably boxed-in) source of light and heat source is placed directly above the sample causing the material to dry out. Animals migrate downwards through the sample. Rapid drying should be avoided as this will kill smaller organisms before they escape from the sample. Simple extraction funnels can be readily improvised in the field. The very simplest apparatus using a large plastic sieve, a bit of 3mm metal mesh

and a bung is illustrated by Lincoln and Sheals (1979).

An excellent piece of equipment for extracting invertebrates from moss, dead wood, litter and even things like bird nests in the field, is the Winkler bag. The bags are commercially available (see page 85) but a full account on the use and construction of this lightweight and highly effective apparatus is given by Owen (1987). The Winkler bag is relatively cheap to make and, unlike other devices, does not require an artificial heat source to make it work. In essence a wire-braced, box-shaped (although I can see no reason why it could not be circular), calico bag is used to enclose measured samples of debris contained inside coarse netting. The bottom section of the bag tapers to a collecting bottle, which may be used dry or with alcohol. The top of the bag is tied up and the whole thing is hung up under cover to dry out gradually. Animals from samples which start fairly dry will be extracted in a few days, but wet samples may take more than a fortnight. To speed up the process it is advisable to occasionally take the samples from the inner bags and mix them to redistribute the wet bits. Winkler bags are particularly good at collecting small beetles, ants and cockroaches.

Extraction methods, such as Baermann funnels, are suitable for the extraction of very small or delicate organisms from soil and other fine grained samples, however, unless you are lucky enough to have a fully equipped research station nearby, specialised devices such as the Macfadyen-Tullgren funnel (Macfadyen, 1962) or Kempson's apparatus (Adis, 1987) are rather complex, expensive and not really appropriate for expedition or field use.

4.13 Aquatic sampling techniques

Chapter 6 in *Ecological Methods* (Southwood, 1978) is compulsory reading for any expedition dealing with insects in freshwater habitats.

In the main, insects can be simply collected from freshwater habitats using any number of hand net designs (fig.23).



Figure 23: A simple water bug catcher made by taping a kitchen sieve to a length of wood and a professional pond net

Catches are simply tipped into white sorting trays or through sieves of various mesh sizes (0.5mm or greater is fine for insects and larvae) and the species required removed for preservation in alcohol. Hand nets are long-handled and for deep rivers or lakes should be able to take additional (screw-on) handle sections. The net bag should be made from strong polyester or 1mm nylon mesh. The most common hand nets have circular or D-shaped mouths. Insects are caught by passing the net through the water or vegetation. Hand nets can also be used downstream to collect anything dislodged from vegetation or stones. Drift nets are rectangular-mouthed, long, tapering nets which are staked or weighted down to collect organisms from shallow streams. A Surber sampler is a drift net with side screens fitted to a quadrat of known area (usually $0.1m^2$) which can be used to sample from shallow streams where the flow is less than 10cms/sec. Stones and plants within the quadrat are kicked, jarred or brushed and the organisms dislodged are carried into the net by the current.

While it is very easy to collect insects in a relative manner from aquatic habitats, difficulties occur when trying to take reliable samples from a known

volume or area of habitat. When collecting from open water you can easily calculate what volume of water has been sampled by your net. A number of sampling cages, cylinders, and grabs have been made which enclose a known volume of vegetation, bottom substrate or sediment (Southwood, 1978). In deeper water, dredges and trawls can be used to sample a known unit of substrate and open water habitat respectively.

Emergence traps, either fixed to the bottom or floating on the surface, can be used to collect insects that emerge as adults from freshwater habitats. Two types are described by Williams and Feltmate (1992). Various sorts of artificial substrate (plastic mesh, discs etc.) or natural substrate (leaf packs, stones) have been used to encourage colonisation by invertebrates. These types of sampler are left in place for a specified time and then retrieved.

Good quality hand nets and other aquatic sampling equipment can be obtained from GB nets (see suppliers: page 84).

4.14 Taxa specific techniques

There are a number of useful field techniques, mainly specific to particular taxa, which do not easily fit into the categories already mentioned.

4.14.1 Fleas

Fleas (Siphonaptera) are specialised ectoparasites of mammals and birds. Unlike parasitic lice (Phthiraptera), they do not spent their entire life attached to the hosts fur or feathers. Flea eggs are shed in the nest or lair of the host and the larvae are free-living, eating organic debris and the droppings of the adult fleas. Fleas and other ectoparasitic insects, such as lice and some Diptera, may be obtained by searching, combing or fumigation (using chloroform or ether) of live or dead hosts (Southwood, 1978). Remember to find out about any legislation in this area. You may need licences to trap and handle live birds or mammals and it is essential to get training, especially if the use of anaesthesia is planned. Much larger numbers of fleas and other associated flies and beetles can be found in nests and lairs. Nests and lairs may be located visually but mammals might only be active after dark or escape through vegetation after being released from live traps. An inexpensive spool-and-line technique for following small mammals to their nests is described by Boonstra and Craine (1986). Essentially a thin thread is attached to a live trapped animals usually by gluing it to the fur. When released, the animal pulls thread from a spool which allows the researcher to locate the nest. Early designs allowed tracking up to 200 metres but improvements have increased this to more than 1,500 metres, depending on

vegetation. Hawkins and Macdonald (1992) evaluate the technique for use with larger mammals.

Invertebrates living in nest material can be extracted using Berlese/ Tullgren funnels or Winkler bags. These methods will also yield flea larvae which would not be present on the body of the host. I am grateful to R.S. George for the following description of his technique for collecting fleas from nest material. The equipment needed is a large, steep-sided bowl, specimen tubes containing 70% alcohol, a dissecting needle and a large sheet of white paper. Before examination, nests must be kept separately in sealed boxes, tins or polythene bags. The collector should wear a white shirt and work with rolled-up sleeves. A small amount of the nest material is placed in the bowl which should stand in the middle of the white sheet. Adult fleas are picked up on the end of the alcohol-moistened, dissecting needle and transferred to a tube. The material will also contain flea cocoons. These can be opened at the truncated end and, usually, adult fleas will emerge with a rush only to be collected. When no more adults can be found the material can be transferred to a sealed box either to breed out any remaining larvae or to allow a re-examination. Any fleas jumping free from the bowl are easily seen against the white paper, shirt or bare arms and collected. This technique will produce many times more specimens than funnel extractions and is essential if you need quantitative data.

4.14.2 Butterflies

If you can recognise butterfly species you encounter you need not kill any (see Conservation: page 20). However the butterfly transect walk techniques described by Pollard and Yates (1993) need accurate identifications. This might be possible in Britain but will be much more difficult in Borneo or Belize. Butterfly field guides are available and in some cases you can make up your own field identification guide based on a variety of sources, expert advice and existing museum collections. Mark-capture-recapture techniques to obtain absolute population estimates are described by Southwood (1978).

4.14.3 Bees

Bees can be most easily collected at flowers but try looking for nest sites in south or south-west facing exposures of bare earth or sand. In savanna and deserts some species will often nest in the vertical faces of dried up river beds. These and other species of bees that use the pith-filled cavities of plant stems, abandoned wood boring beetle burrows and empty snail shells can be trapped in artificial nests. Trap nests can take the form of bundles of bamboo, pithy plant stems or even wide drinks straws tied together or blocks of softwood drilled along the grain using a variety of holes sizes from 2

millimetres to 2 centimetres in diameter. If appropriate trap nest designs are used, bees will start to colonise within hours of them being positioned in the field and occupancy rates can be as high as 50-70%. For the construction of trap nests see Krombein (1967). O'Toole & Raw (1991) give outlines of the biology of various sorts of cavity-nesting bee species.

4.14.4 Termites

In tropical habitats termites are vitally important as decomposers and soil conditioners and are significant pests. There are also many interesting and largely unstudied species who live as inquilines in termite colonies. Numerically abundant (up to 10,000 individuals per m^2) the biomass of termites may outweigh all herbivorous vertebrate animal species put together. Despite this, termites do not appear a great deal in samples collected using standard entomological techniques. Even insecticidal techniques will not sample termites nesting in tree canopies. If sprayed or fogged they will simply retreat inside their tunnels and nests. Sampling obvious structures, such as mounds, will provide only a tenth of the termite abundance in some habitats. Beware of snakes resting in termite mounds and resist the urge to thrust your arm down a ventilation shaft to "see what might be inside." Eggleton and Bignell (1995) consider the various strengths and weaknesses of a variety of termite sampling procedures and develop a standard, stratified sampling protocol designed to take into account the patchy distribution and specialisation of termite assemblages.

4.14.4 Tsetse flies

Anyone particularly interested in studying horse flies, black flies, tsetse flies and other blood feeding species should consult chapters 7 & 8 in Muirhead-Thomson (1991). Of particular interest is the biconical Tsetse fly trap developed by Challier and Laveissiere (1973). This highly efficient design is reasonably simple to make and can trap most of the tsetse flies in a particular location (see fig.24).



Figure 24: A biconical tsetse trap

4.15 Canopy techniques

All that is known to date about collecting arthropods from forest canopies can be found in two books: *Forest Canopies* (Lowman and Nadkarni, 1995) and *Canopy Arthropods* (Stork, Didham and Adis, 1996). Of particular interest to expeditionaries is Chapter I (Moffett and Lowman) in *Forest Canopies* which discusses canopy access techniques and Chapter 5 (Erwin) which deals with measuring arthropod diversity in the tropical forest canopy. The first four chapters (various authors) of *Canopy Arthropods* cover all aspects of canopy sampling. In short these two books, which have extensive bibliographies, provide up-to-date information on the biology of canopy arthropods and how to sample and study them.



Figure 25: Collecting trays under a flowering Acacia, Senegal

Essentially you can either go up to collect material using a variety of approaches ranging from climbing ladders and cranes to net rafts placed on the canopy by means of a dirigible (airship, balloon) or you can get the material to fall down using insecticides. Somewhere between these two extremes are things like composite traps which are hoisted into the canopy (Basset, 1988).



Figure 26: Mist-blowing low savanna tree canopies

The variety of insecticidal knockdown techniques that have been used in various parts of the world have a few things in common. Collecting trays of a known area are set up on the ground below or suspended from vegetation below the tree to be sampled. The trays, which can be conical or pyramidal, are best made from smooth, close woven material such as rip-stop nylon (fig.25). They should ideally have an integral collecting bottle at the bottom to speed up collection and minimise the loss of material. Under small trees or

bushes, large plastic washing-up bowls can be used. A fast-acting insecticide such as a synthetic pyrethroid is blown into the foliage in the form of a fine mist or as a fog (fig.26). Misting or fogging is commonly done early in the morning when there is no wind. Obviously the longer the drop the greater is the risk that a gust of wind might blow small insects clear of the trays. Wide treatment of the canopy around the area delimited by the trays below will reduce this error. Early samples are also thought to be more representative of the canopy fauna (Stork and Hammond, Chapter 1 in Stork *et al*. 1997). Drop time is an important factor. Most studies have shown that 60-90 minutes is more than adequate. Samples should be preserved in alcohol as soon as is practicable and can be sorted roughly and separated from debris in the field back at base (fig.27).



Figure 27: A minute portion of a cleaned-up tree canopy sample in alcohol

As you would predict, some techniques are better for some things than others. Hand searching in canopy foliage tends to miss small insects whereas insecticidal fogging or mist-blowing tends to under-record larger specimens. Other problems with canopy sampling are that although there have been a number of well documented studies, they have used a wide range of sampling methods. Fogging, mist blowing, fumigation and gassing are not comparable and the use of different insecticides, even different formulations of the same

insecticide, have added to difficulties (Erwin in Lowman and Nadkarni, 1995). The take-home message here is: be aware of the many difficulties in sampling from canopies and select appropriate sampling techniques (see Basset *et al*, Chapter 2 in Stork *et al*., 1997).

* It should be noted that a MAFF licence is needed to operate an insecticide mist-blower or fogger in the UK. Similar rules might apply in other countries so check before you go.

4.17 Collecting in caves

For those of you who wish to combine speleology with biology the study and characterisation of cave faunas is still a fresh field. In the past, the fauna of limestone cave systems in Europe and North America have received much more attention than those of the tropics (see Vandel, 1965; Howarth, 1983 and Chapman, 1993). However, in the last 20 years it had become clear that tropical caves are as rich if not richer (see Deharveng & Leclerc, 1989; Ashmole *et. al*., 1992). Representatives of large numbers of new troglobiont taxa have been described and it is certain that only a small proportion of the total has been discovered.

Collecting methods are varied but generally include pitfalls traps, baited bottle traps and timed or fixed-effort hand searching (Ashmole *et. al*., 1992). These techniques can be modified to suite the habitat but a typical sampling protocol might involve sampling at a number of sites to give a certain number of trap-days at each site.

4.17.1 Pitfall traps:

Straight-sided, screw-topped polyethylene plastic jars are carefully dug into muddy, sandy or gravely substrates with their tops flush to the surface. The jars are then filled with approx. 50ml. of modified Turquin's liquid. The liquid is made up from 10g chloral hydrate, 5ml formalin, 5ml glacial acetic acid and 1ml of detergent made up to 1 litre with a dark (preferable), local beer. Ten pitfall traps could be used at each site and, left undisturbed for three days, will give 30 trap days. Specimens trapped are sieved from the traps using a fine tea strainer and stored in 70% alcohol.

4.17.2 Bottle traps:

Straight-sided, wide-necked, screw-topped polyethylene plastic bottles having a capacity of about 250ml (not critical as long as you are consistent) are wedged against rock surfaces and in crevices at an angle of about 45^o. The bottles are one third filled (approx. 75ml) with modified Turquin’s liquid

further baited with a small piece of ripe "Danish Blue" cheese. The cheese should be kept above the level of the liquid by pressing it into an upturned specimen vial cap, through which a 60mm long galvanised nail has been hammered. As for the pitfalls, bottles traps are left in situ for three days before the catch is sieved out and stored in 70% alcohol.

4.17.3 Hand searching:

To complement the trapping at each site (define an appropriate area in m^2 around the traps) there should be a constant number of man-hours hand searching. Any animal encountered can be collected using fine forceps or a pooter and transferred directly into a vial containing 70% alcohol. Care should be taken to examine every possible microhabitat, such as rotten wood, under stones, and in rock cracks and crevices present within each area.

4.18 Collecting from live animals

Blood feeding and other sorts of insect may be collected using live animals of various sorts, including humans, as bait (Southwood, 1978; Muirhead-Thomson, 1991). There are a number of obvious hazards. Large animals, even domesticated ones, can be dangerous at both ends. Tethered or not, approach from the front. Flies can be caught directly as they feed but only a very tame animal will remain calm while an excited entomologist swipes at its hind quarters with a net. Small animals, such as rodents and bats, can give painful bites and may carry diseases such as rabies. As always, assess the risks of what you propose to do and seek expert guidance. Always get training in handling live animals and wear protective clothing if required. There is a species of crab louse, *Pthirus gorillae* (Phthiraptera: Pthiridae), that lives in the pubic hairs of gorillas. As far as I know, no-one has yet attempted an ecological study of these interesting parasites.

Parasitic lice can be picked or combed from the fur or feathers of mammals of birds caught in live traps or mist nets. Another way might be to fumigate the body of the animals with carbon dioxide, ether or a fast acting insecticidal powder or spray but anyone who has tried to treat even a moderately feisty domestic cat will know that both parties can finish up distressed. Southwood (1978) reviews a variety of methods for sampling from live hosts but it is clear that absolute counts of many ectoparasitic species can really only be obtained if the host is sacrificed so that the fur or feathers can be subject to detailed examination.

4.18 Making your own equipment

There is a tendency to assume that collecting equipment cannot be much good if it is not expensive and bought from a proper scientific supply company. In many instances this is simply not true and, in any case, commercially available products are based directly on home-made designs gleaned from the publications of professional and amateur researchers. There may not be a piece of collecting gear that does what you want and you may have no alternative but to make something yourself. It can be very enjoyable and can produce very interesting results. My favourite bit of kit for catching surface-living water bugs is a kitchen sieve taped to a length of stick (fig.23). The most commonly used pitfall traps are disposable drinks cups. You can save a great deal of money by using your imagination. This is not to say that making any particular piece of collecting equipment will be straightforward. Your idea must made into a prototype and then field-tested to see that it does what it is supposed to do. That trap designs should be tested scientifically has been amply illustrated by Phillips and Wyatt (1992). The authors wanted to understand why similar pest monitoring traps caught very different numbers of cockroaches. What they discovered was that the angle of the entry ramp to the trap box was critical. An entry ramp of 30° was found to be best and caught far more than either 60° or 0° ramps. The steep and flat ramps both caught a similar, lower number of cockroaches but for different reasons. Observation showed that while fewer cockroaches went up the steep ramp, none escaped whereas of the many cockroaches that entered the trap by the easy, flat ramp, half escaped.

Once you have designed and tested your equipment it is important that it is properly used and located in the field. Many studies have shown that all manner of variables, such as height, orientation and position are important factors in trap efficiency. For a review in relation to trapping flying insects see Muirhead-Thomson (1991).

Some very useful specialist items are probably best home made although it might be possible to find a supplier somewhere. One such piece of kit is a D-shaped net with a short, squat handle for collecting insects from under the bark of trees and similar habitats. The front section of the net (the straight bit of the D) is made from flexible plastic tubing or metal strip to allow the net to be pressed firmly around the curvature of the tree trunk. The short handle is braced against your body to free both hands for picking and brushing the bark. Dislodged insects fall directly into the net below.

4.19 Data recording

The recording of data in the field is one of the most important aspects of any field trip and is often done badly. Always record your field notes in a bound notebook and never in loose leaf folders or binders. Pages fall out and get lost and it is always the most important pages. Pocket sized notebooks are best and special notebooks with water proof paper (AquaScribe™) are essential for writing in the rain or in permanently wet habitats, such as swamps and caves.

Specimens collected must have adequate data associated with them. Information on where, when, who and how any particular insect was caught should be as full as practicable. For instance “Tanzania, August 1996” is clearly inadequate as compared to “collected at UV light, 12th August 1996, Ibaya Camp, Mkomazi Game Reserve, Tanzania, G.C. McGavin.” In the field the data should be kept closely with the specimens (attached to it or inside the tube or envelope). Access to GPS (global positioning systems) enables collecting sites to be pinpointed as never before.

Code numbers kept with the specimens which refer to fuller data in a notebook may seem easier but, if the notebook goes astray, all is lost as the material is useless without data. Do not write on the outside of tubes or on tube tops. Tube tops can get accidentally switched, labels can fall off or get rubbed off. Labels for putting inside tubes of material stored in alcohol is best done on good quality paper with pencil or waterproof drawing ink. Allow ink labels to dry thoroughly before use. If you are collecting a large number of samples or if you are working in difficult conditions, the use of pre-printed labels will save a lot of time but be sure to test for alcohol-fastness. Inkjet and laser printed labels will run or the letters will float off in alcohol. I am assured that inkjet cartridges filled with permadri™ ink are alcohol proof (Graphic Utilities, Chesey Grove road, Fort Fairfield, Maine 04742, USA). Good quality photocopies of the data sheets are good but it is still best to test them at home.

Never think that you will recall where or when a particular specimen was caught when you get back from your trip. Remember at all times **that specimens without proper data are completely useless**, a total a waste of your time and effort, and, if you have been fund-raising, a waste of other people’s money.

In the same way that you would back-up valuable computer data on separate disks, duplicates of vital specimens are best kept in separate locations.

Section Five

KILLING METHODS

It is chiefly through the instinct to kill
that man achieves intimacy with the life of nature.

Lord Clark (British Critic) from
The Faber Book of Aphorisms (1964)

The sense of death is most in apprehension,
And the poor beetle that we tread upon,
In corporal sufferance finds a pang as great
As when a giant dies.

William Shakespeare; *Measure for Measure*
III. i . 75

If you are working on the behaviour or ecology of a known species for example there will be no need to kill many specimens, except perhaps to check identifications. Photographic records of larger species, such as butterflies, might be good enough for most purposes but you may still need to take a few specimens as vouchers. For most insect field work however, especially in regions where the fauna is relatively unknown, you will not be able to do anything of value without killing.

I'm afraid there are no really nice ways to kill animals, insects included, so if you have serious worries of any kind about this, think of something else to do on your expedition. There are no clear guidelines about what constitutes the best or most humane way to kill insects but common sense should tell you that if insects are sentient organisms none of them are going to be much fun. The quicker the better is a good principle as long as the specimens are intact and useable at the end (see Martin, 1977; Arnett, 1985 and Davies and Stork, 1996).

More detailed information on the chemicals mentioned in this chapter and elsewhere in this book can be found in a number of technical publications of which the Merck Index is widely available (Budavari, 1996). When using chemicals thoroughly familiarise yourself with the hazards and precautions. Appropriate safety procedures must be observed at all times and bottles and jars should be labelled correctly.

A traditional method to kill insects is the cyanide killing jar. The technique is still favoured by some professional lepidopterists and orthopterists as it kills more quickly than other methods, keep the specimens dry and does not cause colour changes (ethyl acetate fumes, for instance, can cause some green pigments to fade). Glass, or preferably high density polyethylene jars for field use, are made in advance and with careful preparation may give many months of service (Arnett, 1985; Townes, 1973). A typical jar uses a packed, 5mm layer of powdered potassium cyanide crystals under a 1cm thick layer of sawdust and topped with a 1cm layer of plaster of Paris. The plaster should be allowed to dry out thoroughly before the jar is ready for use. Townes recommends drying in an oven but care should be taken to ventilate the room and vacate it while drying proceeds. In museums cyanide jars are made under fume hoods. In its dry state a cyanide jar will have a shelf-life of one or two years. Once in the field a few drops of water soaked into the plaster layer will react with the potassium cyanide to produce cyanide gas. Full production of the gas will build up over a day or two and should keep going for many weeks. Contaminated equipment should be immersed in dilute sodium hydroxide solution to which is added an excess of ferrous sulphate solution. After an hour rinse and flush away with an excess of water.

Butterflies caught in a net can be quickly incapacitated by a gentle pinch across the thorax using the nails of the thumb and forefinger. The technique renders wings useless and the specimens can then be transferred to a paper envelope without the risk of them flapping around and losing wing scales. The envelopes can be stored flat in a tupperware™ container temporarily before being killed in an ethyl acetate jar. Pinching does not work very well on moths.

When using a malaise trap the catch is generally going to be killed and stored in alcohol anyway. For many other mass collection techniques, such as suction sampling or sweeping (the catch will be live) or mist blowing (the catch will already be dead), the sheer volume of material means that that simple immersion in alcohol is the only practicable combined killing and storage method.

Do not forget that if you are collecting particular taxa or species you can separate these from your sweep or suction sample manually and release the rest into surrounding vegetation. Fragile insects collected in a pooter will die quite quickly if left in direct sunlight.

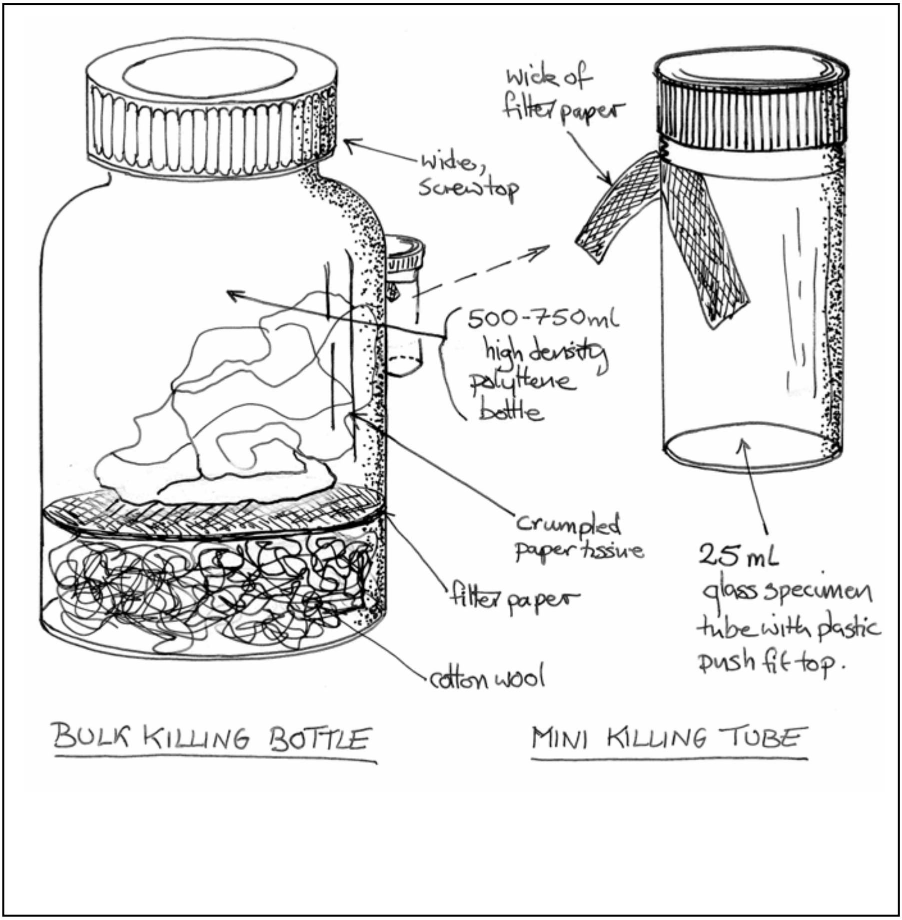


Figure 28: Killing bottles

Very large insects, such as dung beetles, can be killed very quickly by the injection of a small quantity of saturated oxalic acid solution into the thorax or abdomen through the intersegmental membranes. But take care, apart from being poisonous, oxalic acid is a white, crystalline powder which might arouse suspicion at customs. Dung beetles and other fairly robust species can also be killed very quickly by dropping them into boiling water.

The vapours of various chemicals, such as benzene, carbon tetrachloride, ether and ammonia have been used in the past to kill insects. Many of them

are dangerous and some highly inflammable but probably the safest alternative to cyanide in the field is ethyl acetate. The fumes of ethyl acetate kill more slowly than cyanide and are still flammable. Killing bottles or tubes can be made with a layer of plaster of Paris at the bottom onto which a few drops of ethyl acetate can be dropped. More simply, a layer of cotton wool with a few pieces of filter paper on top can be arranged inside a polythene bottle (fig.28). Bulk killing bottles should have a capacity of at least 500ml, and a wide top to allow you to get large specimens inside easily. Specimen tubes of strong glass or polythene are best for killing small insects. A wick of filter paper with a drop of ethyl acetate is held in place by the push fit top. Glass tubes are acceptable here and have the advantage of being transparent. It is best to have a small wad of tissue paper inside the killing chamber to soak up excess moisture. Ethyl acetate and acetone (sometimes used as a substitute and widely available as nail varnish remover) will dissolve styrene plastics. When dead, the insect should be removed and pinned or stored as required (see Specimen Preservation: page 75).

If you have no chemicals but have access to a freezer you can kill insects by freezing them in tubes. Do not open the tubes until their contents have reached ambient temperature or water vapour will condense on the surface of the specimens and mould will develop rapidly.

Section Six

SPECIMEN PRESERVATION

"Alas! - those who in museums contemplate the works of nature do not think of all the perseverance, trouble and anxiety required before they are safely brought home."

H. Mouhot, three months before his death from jungle-fever on the River Mekon, 1861.

6.1 Field preservation

Once having collected your insects and killed them in an appropriate manner the next step is to get them home safely. If working on particular taxa consult an expert in the group before you go. Stehr (1978) describes techniques for the preservation of immature insects. In the field, the simplest techniques are drying or storing in alcohol. Most robust insects, such as beetles and bugs, can be simply dried in air and sorted in layers of cellulose wadding or tissue air dried but delicate or soft-bodied species and spiders should be preserved in alcohol. Some coleopterists preserve small beetles in a 4% solution of glacial acetic acid until they are dried and set. If kept cool the material should be fine for many months. Moths and butterflies should never be stored in alcohol and are best dried in small paper envelopes or folded paper triangles or set and pinned directly into boxes. The final setting and pinning of insects in the field is not good if you envisage collecting any more than a few specimens. Set specimens take up a lot of room and are difficult to transport safely.

A problem can arise with large, fat-bodied species, such as some grasshoppers and mantids. The large quantity of internal tissue can take forever to dry out properly and decay often sets in. A good technique is to stuff the abdominal and thoracic cavity with cotton wool. This is not as difficult as it may sound. Using a pair of fine dissection scissors make a slit in the ventral surface of the abdomen. You do not need to open the abdomen up from stem to stern. Remove the abdominal contents with a pair of fine watchmakers forceps. Be sure to reach up into the thoracic cavity to remove the front portion of the gut. Do not rip the guts from the rear end of the animal as you may damage or remove parts of the genitalia which may be of value in identification at a later date. Use your scissors to cut through the rectum as close to end of the abdomen as you can easily go. You do not have

to remove every shred of soft material from the inside. Starting at the thoracic end use your forceps to push little bits of cotton wool into the cavity. Do not overstuff. When finished, the abdomen should be as close to the shape it is in real life. You do not need to sew up the incision as the cuticle will dry in position without being held in place.

The most useful field preservation technique for adult insects that I and many others have successfully used for years involves the use of chlorocresol (more correctly 4-Chloro-m-cresol) (Tindale, 1961). A small quantity (a level teaspoonful) is sprinkled in the bottom of an airtight plastic food storage box and covered with a layer of tissue or wadding. On top of this, specimens in labelled paper envelopes or layered in wadding or tissue are placed. A few drops of water are sometimes advised to keep the whole contents of the box slightly damp but I have never found the need for this as the moisture from the specimens is generally sufficient. Every tenth layer or so, more crystals of chlorocresol can be sprinkled on top. A crumpled tissue or two keeps the layers in place until the box is full, when a final generous sprinkle of cholorcresol should be added. The chlorocresol acts as a fungicide and bactericide and will prevent mould and decay setting in. The insects will keep moist and relaxed for months so that when you come to mount them the job is very easy. Air dried insects must be first relaxed in very warm humid conditions before they can be pinned and have their appendages arranged. The chlorocresol technique avoids all this and even butterflies and moths treated in this way will remain “settable” and in good condition for a long time.

Despite the fact that aeroplanes are stuffed full of aviation fuel and carry large quantities of flammable alcohol for human consumption, carriers are not keen on passengers packing bottles (plastic or otherwise) of ethyl alcohol in their baggage. In the nineteenth century, collectors and explorers often sent biological material home preserved in brandy or rum but this practice is not ideal today for a variety of reasons. Expense apart, the alcohol content is not high enough. Most spirits are between 30-60% alcohol whereas soft-bodied adult insect specimens, immature insects or spiders require preservation in 70% - 80% alcohol. For field preservation methanol will do as well as ethanol but, as it is slightly more toxic and volatile and has the, more corrosive, formic acid, not acetic acid, as its final oxidation product, it is not best for longer term storage. If you cannot get diluted ethyl alcohol (70-75% is common) you will have to a dilute stronger alcohol. Sometimes what is sold as absolute alcohol is not 100% but actually nearer 95%. Dilute to the required strength with water (it is not necessary to use distilled water as a diluent, clean rain water or filtered water will do fine).

Attempting to carry any more than a couple of hundred millilitres of alcohol in airline hand or cabin baggage is not really recommended. There is the risk of leaks or confiscation, neither of which would be a good start to your expedition. I have experienced no great problems in obtaining ethyl alcohol on field trips although it is sometimes necessary to spend a few days in a large city to track it down in chemist shops or scientific suppliers. This sort of pre-field activity can be combined with other official duties, such as visiting appropriate authorities to get last minute permits and clearances, as well as just out of courtesy. In many countries, 70% isopropyl alcohol (2-propanol) is widely available as rubbing alcohol. This is fine for temporary storage but should be changed for ethyl alcohol when you get home.

On the return journey, when your specimen tubes are filled with preserved insects, officials no longer take any interest in the alcohol but may want to know what the specimens are, so make sure you have all the relevant export permits. The golden rules when storing and transporting insects in tubes of alcohol are to fill the tubes right to the top but never fill the tube more than half full with specimens. You may have to carry more tubes home but if you do not follow this rule the specimens themselves dilute the alcohol and the strength of it may fall below 40% at which point things will start to rot. A simple loose tissue or cotton wool plug gently pushed on top of the specimens will keep them from being shaken about in transit. As a general rule I always take material home in my hand baggage. Items such as camera equipment can be easily replaced and should be consigned to the hold.

If you are collecting material for molecular systematic procedures or other studies that require the extraction of DNA you will need to seek specialist advice. Ideally, material should be preserved in liquid nitrogen but this might be difficult in many field locations. A good alternative is storage in 100% ethyl alcohol (absolute ethanol). Samples should be stored in a freezer as soon as possible. Be very careful of contamination and always wear a fresh pair of surgical gloves when handling material.

6.2 Long term preservation

For some studies involving large amounts of material it might be simplest to leave specimens in alcohol until such times as they really need to be prepared in a more specific manner. Spirit-preserved material does, however, require maintenance. For long term storage, critical specimens should, ideally, be kept in a freezer. This will reduce the need for topping up the alcohol and, as an extra bonus, keeps internal organs in absolutely first class condition for decades.

Usually identification and systematic study of specimens will require them to be mounted dry. Pinning insects enables then to be handled, examined and stored safely.

Specialist taxonomists have particular ways of mounting their specimens and, if you are expecting them to work on your material, it would be good to know how they would like the material presented. I was once given a soggy cardboard box filled with half-empty tubes of alcohol containing a mush of partially decayed, unlabelled insect material. This material was the sum total of three months collecting by ten young people who had, sadly, paid good money to be part of a “scientific” expedition to a very interesting part of Africa. The box, and its bearer, were sent packing, the smell lingered in my office for days. I often wonder if the collectors ever found out just how purposeless their misdirected efforts had been. The organisers had broken practically every rule of field work there is to break.

Sometimes inappropriate mounting can destroy the very features required for proper identification. General insect mounting, preservation and curatorial techniques can be gleaned from a variety of sources but among the best are Martin (1977) and Walker and Crosby (1988). Carter and Walker (1998) deal with all aspects of the physical care of botanical and zoological collections and give much practical guidance over the whole field of natural history curation. Publications giving specialist techniques for particular orders of insects, such as the Isoptera (Krishna and Weesner, 1970), Thysanoptera (Lewis, 1973) the Hemiptera (Dolling, 1991; McGavin, 1993), Coleoptera (Cooter, 1991), Diptera (Stubbs and Chandler, 1978), Lepidoptera (Dickson, 1992) Hymenoptera (Gauld and Bolton, 1988), Plecoptera, Ephemeroptera and Trichoptera (Macan, 1982) should be consulted, where appropriate. Many of these books also give details of collecting methods.

Most robust-bodied specimens can be simply taken out of alcohol, dried and then mounted in the manner appropriate to the taxon concerned but small or delicate specimens will need some treatment or they will shrivel up and become useless. There are some simple techniques that will allow these specimens to be taken out of alcohol, dried and mounted without bits or all of the body collapsing. Critical point drying is the best approach but requires expensive equipment and time. A cheap and effective alternative to critical point drying using acetone has been developed by several researchers. I am grateful to Simon van Noort of the South African Museum in Cape Town for the following recipe which can be used for a range of small or weakly sclerotised insects, especially parasitic wasps.

The quickest and easiest approach, particularly where there are large numbers of specimens, is to treat the whole sample in one go. Pipette the specimens in alcohol on to a square piece of stiffish card, the edges of which are folded up to make a miniature sort of tray. Arrange them in the position required for subsequent mounting at this stage. Place the card in a petri dish lid to keep the specimens saturated in alcohol while getting them into position. The specimens are best on their right hand sides with wings folded flat above the body, which takes a bit of time, but is worth the effort, so that when it comes to mounting it is simply a matter of placing the specimen on a blob of glue on a card point. Once the specimens are arranged, drain off the excess alcohol by lifting up the card and letting it run off before placing the card tray into an acetone saturated environment. Acetone is soaked liberally on to cotton wool at the bottom of a sealable acetone-proof container and the cards, bearing their arranged specimens and still wet through with alcohol, should be placed horizontally or vertically, clear of the acetone, for a minimum of six hours. To dry the material, lift out the card tray, which is now saturated with acetone, and place it in a petri dish under a 60watt desk lamp (close to the bulb) for 20-30 minutes. Once the specimens are dry be careful not to breath too hard over them otherwise they end up all over the place. Once dry they do not need to be mounted straight away and can be kept quite safe in a petri dish in a pest free environment. The method generally gives good results but sometimes specimens may still collapse even though others in the same sample are fine.

Another technique for the recovery of non-robust specimens from alcohol storage is particularly useful for small flies or bugs. Start by pinning the specimens direct from the alcohol. Stand the pinned specimens in a tube filled with 2-ethoxy-ethanol overnight (12 hours minimum). Transfer to a tube (glass) filled with ethyl acetate (1.5 hours for small species - 3 hours for larger species). When the specimen is removed, the wings particularly will stick together or crease up. Dab off excess ethyl acetate with a tissue and blow rapidly on the specimen to release the wings. If the wings do not immediately spring out, tease them out with a fine pin or forceps (J. Ismay, pers. com).

Bees which have been stored in alcohol cannot simply be taken out and dried, as the dense hair covering becomes matted and flat. A near life-like dried specimen can be prepared in the following manner. Remove the bee from the alcohol and dab dry with tissue to get rid of most of the fluid. Immerse in 2-ethoxy-ethanol for 24 hours. Remove excess alcohol by dabbing with tissue. Pin specimen in the normal manner, placing legs and

wings in the desired position. The male genitalia may be exposed at this stage using a small hooked pin. Soak the pinned specimen in ethyl acetate for 24 hours. Remove and dry immediately from behind with a small hair drier. The hairs will fluff up and remain erect (C. O'Toole, pers. com.).

Should important, spirit-preserved material ever dry out it can be rescued by soaking in a 2% solution of trisodium orthophosphate. The rehydration rate depends a great deal on the size and condition of the specimens but it is best to check them every hour or so. A good indication that the process is complete is when the specimens sink to the bottom of the container. The specimens should then be thoroughly washed in distilled water before being put back into 70% alcohol.

6.4 Posting material

In some cases it might be more convenient to send collected material back from the field by post. On your return you may have to post material to experts. There are risks associated with posting material and steps should be taken to minimise them. While sending specimens by airmail and registered or some other kind of insured postal service is essential, proper packing is just as important. The following guidelines should avoid the recipient opening a box of detritus.

Set or mounted specimens should be pinned into a small box lined with cork or polyethylene foam and cross-pinned to ensure that they will not swing around or wiggle loose. Never pin into polystyrene foam as it is brittle and does not grip pins. Never pin a very large specimen, such as a scarab beetle, along with more delicate material. If the large one comes loose in transit it will reduce the rest of the box to dust in short order. The lid of the small box should be secured with rubber bands or tape. The small box should now be placed inside a larger box with at least 10cm clearance all round which is filled with polystyrene chips, crumpled newspaper or other packaging. Do not compress the outer packing too tightly as it will lose its shock absorbing qualities. The box should be wrapped in strong paper, if available, secured with string or parcel tape and addressed on several sides. Also ensure that all necessary customs forms are completed and that the parcel is clearly labelled with the following information:

FRAGILE! CUIDADO! VORSICHT! PRECAUTION!
Dead Insects for Scientific Study
Insectes Desseches pour L'etude Scientifique
Getro. Insekten für Wissenschaftliche Zwek
NO COMMERCIAL VALUE - NO VALOR COMMERCIAL

There are two schools of thought when it comes to posting material in alcohol. One advises that each tube is filled to the top so that there are no air bubbles to buffet the specimens (see section on alcohol above). The other school advises that most of the alcohol is drained off before posting to reduce the weight of the parcel and the risk of leaks if a tube breaks. You should never use glass tubes in the field in any case and try to avoid brittle polystyrene tubes which split readily. It is best to use polyethylene screw-topped tubes and avoid tubes with push tops which can easily pop out. The tubes are then wrapped in bubble wrap or soft tissue and packed as already described for a box of set specimens. Mason (1974) describes a technique for sending spirit material in plastic bags.

Section Seven

SPECIMEN IDENTIFICATION

For the Snark was a Boojum, you see.

Lewis Carroll; Hunting of the Snark
Fit 8. The Vanishing.

The identification of specimens to the level of order and suborder should not be beyond the capabilities of any careful student with access to basic books. More specialist literature should enable most specimens to be assigned to a particular family. Further identification depends on the order in question. For some insect orders, such as the Lepidoptera, there is a wealth of information and keys galore, while for others there may be little taxonomic help available. Some parts of the globe are much better known in terms of their insect fauna than others. For instance, in the U.K., there are approximately 22,000 insect species and the chances of coming across a new one are slight. However every third or fourth species collected in rain forest or savanna habitats might be new to science.

A mass of unidentified material is not of much use and a waste of your time and other people's money. Specialists are very busy people and their time is precious.

If you need the services of specialist taxonomists, establish contact with them at an early stage. Remember, if they do show an interest in your expedition and say they will look at some of your material, they are doing **you** a favour not the other way round. Exchange letters and communicate at regular intervals to establish an agreed time scale and priorities for action. By these simple means it is easy to avoid the situation where preliminary contact is made and the specialist gives some vague expression of interest only to receive, many months or perhaps a year later, a pile of unsorted specimens which "have to be named" by last week. Even if the specialist in question does remember who you are I do not need to spell out what might happen next.

Find taxonomists who really want to be involved with the project or find money at the start to service this important aspect. You should always try to tailor your project to what you can reasonably expect to achieve. It is, of course, entirely possible that you could become an expert in a particular taxon yourself.

Insects are classified in twenty-nine large groups called orders. Some of these, such as the dragonflies or beetles, will be well known, while others will be much less familiar.

For an introduction to the taxonomic literature up to the late 1970's, the reader should consult Hollis (1980). The most comprehensive single publication to date on the class Insecta is CSIRO (1991).

Stehr (1987) gives a comprehensive key to the orders of immature insects and other selected arthropods.

Section Eight

EPILOGUE

If, by the nature of the methods used, you have collected more material than you wanted you must try to make sure it goes somewhere it can be used, even if only as classroom material. What about the collecting equipment? In many cases this should be donated to host institutions. With any luck you will have had a fantastic experience and made a significant scientific contribution but no one will ever know if you do not tell them through reports, papers and other publications. It is not really enough to simply do the minimum to satisfy the requirements of funding bodies. You owe more than this.

Section Nine

EQUIPMENT SUPPLIERS

There is no space here to give a complete list of equipment but key items are listed for each supplier.

United Kingdom:
[Mycological and entomological instruments and apparatus]
Burkard Manufacturing Co. Ltd. Tel- 01923 773143
Woodcock Hill Industrial Estate Fax- 01923 774790
Rickmansworth
Hertfordshire WD3 1PJ

[Pins, nets, light traps and general collecting equipment]
Watkins and Doncaster Tel- 01580 753133
The Naturalists Fax- 01580 754054
P.O. Box 5
Cranbrook
Kent TN18 5EZ

[Malaise traps, flight intercept traps, Owen emergence traps etc.]
R.S. George Tel- 01202 515238
Marris House Nets
54 Richmond Park Avenue
Queen's Park
Bournemouth BH8 9DR

[Pins and sundries]
D.J. & D. Henshaw Tel + Fax - 01992 717663
34 Rounton Road
Waltham Abbey
Essex EN9 3AR

[Biological water sampling equipment, aquatic nets etc.]
GB Nets Tel + Fax - 01422 845365
Gill Baldwin
45 Burnley Road
Todmorden
Lancashire OL14 7BU

Europe:

[A large range of equipment for ecological and entomological collecting and environmental monitoring]
Gerhard F. Behre — Tel - 0228 614799
ecoTech Umwelt-Meβsysteme GmbH — Fax - 0228 614886
Siemensstraβe 8
D-53121 Bonn

[Winkler bags]
Dr Hildegard Winkler
A-1180 Wien, Dittesgasse 11 — Tel - 0043 1 470 4760
Austria — Fax - 0043 1 470 47604

Israel

[Telescopic butterfly net]
Rani Kasher — Tel - 00-972-6-6951229
Israel Insect Information Centre
Mount Hermon Field School
D.N. Galil-Elyou 2
Israel

USA

[Various equipment]
BioQuip Products, Inc. — Tel – 310 667 880
2321 Gladwick Street — Fax – 310 667 8808
Ranch Dominguez — E-mail - bqinfo@bioquip.com
Ca 90220
USA

[Various equipment]
American Biological Supply Company
2405 NW 66th Court, — Tel - 904 377 3299
Gainesville,
FL 32653

[Various equipment]
Ward's Natural Science — Tel – 1585 359 2502
5100 West Henrietta Road, — Fax – 1585 321 9105
Rochester — E mail – international@wardsci.com
New York 14692

Section Ten

SOURCES OF INFORMATION

World of Learning (1997) 47th edition. Europa Publications, London
[Annual edition giving addresses and contact numbers of universities and learned societies in just about every country of the world]

CAB International*
Wallingford
OX10 8DE
Tel - 01491 832111
E-mail enquiries@cabi.org

[* CABI is the parent organisation of the International Institute of Entomology (Tel-0171 584 0067) and the International Institute of Biological Control (Tel-01344 872 999)]

Hope Entomological Collections
Oxford University Museum
of Natural History
Parks Road
Oxford 0X1 3PW
Tel - 01865 272950
Fax - 01865 272970
Web http://www.ashmol.ox.ac.uk/oum/
E mail – info@oum.ox.ac.uk

Linnean Society of London
Burlington House
Piccadilly
London W1V 0LQ
Tel – 020 434 4479
Fax – 0207 287 9364
Web – www.linnean.org/
E-mail john@linnean.demon.com

Natural History Museum
Department of Entomology
Cromwell Road
South Kensington
London SW7 5BD
Tel -0207 942 5000
Web – www.nhm.ac.uk/research-curation/departments/entomology

Royal Entomological Society,
The Mansion House ,
Chiswell Green Lane,
St. Albans,
AL2 3NS
Tel - 0171 584 8361
Fax - 0171 581 8505
E-mail reg@royensoc.demon.co

The Amateur Entomologist's Society
P.O.Box 8774
London SW7 5ZG
E mail – contact@amentsoc.org
Web – www.amentsoc.org/

[The A.E.S. publishes a range of extremely informative and useful leaflets, pamphlets and handbooks. A prospectus and catalogue of these publications is available from the address given above.]

Zoological Society of London
Regent's Park
London NW1 4RY

Tel - 0171 722 3333
E-mail lzmembership@zsl.org
Web - www.zsl.org/

A great deal of useful information can be obtained from a number of world-wide web sites. Good starting points for general or specific entomological investigations include the following:

http://entowww.tamu.edu/
(Texas A & M University)

http://www.biosis.org
(Internet Resource Guide of Zoology (General Entomology) from Biosis)

http://entomology.si.edu/
(National Museum of Natural History, Smithsonian Institution. Department of Entomology)

http://www.ento.csiro.au/
(CSIRO home page)

http://ww.ento.csiro.au/research/natres/anic.htm
(Australian National Insect Collection home page)

http://www.nhm.ac.uk/science/entom/index.html
(The Natural History Museum, London. Entomology Department pages)

http://www.webdirectory.com/
(Environmental Organisation Web Directory)

http://phylogeny.arizona.edu/tree/phylogeny.html
(The Tree of Life home page at the University of Arizona)

http://www.cabi.org/index/htm
(Commonwealth Agriculture Bureau home page)

http://www.linnean.org.uk
(Linnean Society)

http://www.sel.barc.usda.gov
(Systematic Entomology Laboratory, USDA)

Section Eleven

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Wyman, R.L., Barker, C. & Vispo, C. (eds) (1997) International Directory of Field Stations. Organisation of Biological Field Stations. International Committee Office, New York. (loose leaf). E.N. Huyck Preserve and Biological Research Station. PO Box 189, Rensselaerville, NY 12147 USA. Tel & Fax: +1 518 797 3440, email: rlwyman@capital.net